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TOXAPHENE STATUS REPORT

Gordon Guyer, et al

Environmental Protection Agency
Washington, D. C.

November 1971

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TOXAPHENE STATUS REPORT

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beneficial arthropods
insect pollinators
insect predators
parasites
food residues
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TOXAPHENE STATUS REPORT

Special Report
to the
Hazardous Materials Advisory Committee
Environmental Protection Agency

Consultant Group on Toxaphene

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November 1971

1a

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INTRODUCTION

USE PATTERNS

The amount of polychloroterpene insecticide used in the United States during the past 25 years totals about 940 million pounds, averaging 38 million pounds annually. These figures, based on manufacturer production and sales information, are compared below with more detailed knowledge developed in government studies of farm use of pesticides.

USDA Pesticide Use Census

The Economic Research Service of USDA conducted the first national census of pesticide use in 1965. Data from that study were released in AES Report No. 131, January, 1968. A follow-up study was conducted in 1967, and reported (AES Report No. 179) in April, 1970. In each survey, approximately 10,000 farmers were interviewed in depth to obtain the details of their use of pesticides. Data concerning toxaphene have been abstracted from these reports and are tabulated below. Because the crop designations are not identical in the 2 studies, certain details may not be directly compared, but the major uses and amounts used are clearly set forth.

The data indicate that crop uses for toxaphene (and toxaphene-strobane in 1964) were in the range of 34 ± 3 million pounds annually (Table 1). Livestock use was 4.2 ± 0.5 million pounds. Combined usage was in the range of 38 ± 4 million pounds annually. These figures are only for farm use, and do not include amounts used for any government programs. Such usage would add only modest amounts to the total, however, and the totals do agree reasonably well with figures cited above, which are projected from production and estimated consumption based on sales to formulators. In this regard, it should be recognized that the major producer, Hercules Incorporated, does not market any toxaphene formulations, but sells to formulators who blend, distribute and sell finished formulations. Toxaphene is sold by Hercules Incorporated as technical toxaphene (100%), a 90% solution in xylene, and as a 40% dust base. The 90% solution is preferred by many formulators because it eliminates the inconvenience and expense of handling, melting, and dissolving the waxy solid. The solution is commonly bulk-shipped in rail and truck tank cars.

Certain regional discrepancies have been noted in the USDA data (Table 2). In the Pacific states, for example, the volume of toxaphene used is apparently understated. Major uses in the Pacific states are on cotton, vegetables, and alfalfa seed. California state data for 1970 (from that state's first year of dealer sales reporting) show a total of 2.7 million pounds of toxaphene used on 0.75 million acres. Thus, the USDA data may understate toxaphene usage for that region, but the total usage figures are believed to be reasonably accurate.

TABLE 1

Farm Use of Toxaphene -- 1964 and 1966 Crop Years

Crop Use	Millions of Pounds in Indicated Years 1966	 1964
Corn	0.004	0.1
Cotton	27.3	26.9
Soybeans	1.0	1.3
Tobacco	0.2	0.3
Other field crops	1.6	4.3
Vegetables	0.8	1.2
Total on Crops	30.9	34.2
		2.7 (Strobane)
	Total on Crops	36.9
Livestock Use		
Cattle	3.3	4.3
Swine	0.3	0.3
Poultry	.02	0.00
Sheep	.05	0.1
Other	0.01	-
Total on Livestock	3.7	4.7
Total Crop + Livestock	34.6	41.6

Table 2

Regional Patterns of

Toxaphene Farm Use -- 1964/1966

Region	Millions of Pounds 1966	Millions of Pounds 1964	Millions of Acres Treated (a)(b) 1966	Millions of Acres Treated (a)(b) 1964
Northeast	0.004	.003	.002	.02
Lake States	0.1	.05	.01	.05
Corn Belt	0.4	1.3	0.3	0.9
Northern Plains	0.01	.001	.01	.002
Appalachia	2.5	4.2	0.5	1.1
Southeast	13.7	11.5	1.6	1.8
Delta States	7.2	10.3	1.2	2.2
Southern Plains	5.0	5.1	1.3	1.4
Mountain	1.4	1.0	0.2	0.2
Pacific	0.6	0.8	0.2	0.2
Total	30.9	34.2	5.4	8.0
All Insecticide Use -- 1964/1966				
All organochlorine	82.8	89.8	35.0	41.0
All organophosphorus	36.6	30.5	26.0	24.0
All carbamate	12.4	15.4	5.0	5.0
All insecticides	138.0	143.0	67.0	71.0

(a) Acres treated do not distinguish between an acre treated only once in the crop year or an acre treated many times.

(b) The same acre treated with 3 different insecticides is counted once for each pesticide -- e.g., one acre treated with tox-DDT-methyl parathion would be tabulated as 2 organochlorine and one organophosphorus acres.

Use Outside U.S.A.

Use of toxaphene outside the United States is principally on cotton and livestock, with a variety of smaller uses on vegetables, small grain, peanuts, soybeans, bananas, and pineapple. Toxaphene manufacturing plants are located in Nicaragua and Mexico with local ownership predominating in Mexico. Russia is believed to have toxaphene production facilities, but little is known about the amount and nature of the material produced. Other chlorinated terpene materials are encountered on the world market. One of these, called "Melipax," is made in East Germany. Other so-called chlorinated camphenes have been encountered in Asia. In general, these other "chlorinated terpene" products have been found to be of poor or highly variable quality, and in bioassay tests often require doses several times that of toxaphene to achieve equal insect kill.

While the overseas market for toxaphene is appreciably less than that in the United States, it is also complicated by factors other than safety and effectiveness of the product. Currency and import restrictions, devaluations, trade policy agreements and other complications can make pesticide marketing overseas more difficult than in the United States; and product sales can vary significantly from year to year. Informed opinion concerning the likely impact on the international market of United States restrictions on domestic use of toxaphene is that further United

States restrictions would eliminate much of the use of toxaphene overseas.

Crop Use Outside U.S.A.

Geographic Area	Principal Crop Use
Central America	cotton
South America	cotton, small grains, soybean, bananas
Africa	cotton, vegetables
Europe	cotton, rapeseed, vegetables
Asia	cotton, peanuts, vegetables, rice
Oceania	cotton

Livestock Use Outside U.S.A.

Africa

East, Central and South Africa, including Uganda, Kenya, Tanzania, Rhodesia, Angola, Nigeria and South Africa

South America

- Brazil
- Peru
- Ecuador
- Columbia
- Venezuela

Central America

- Mexico
- Costa Rica
- El Salvador
- Panama

North America

- Canada

None of the countries listed, except Canada, has formally established residue tolerances in meat, although many have noted the 7 ppm tolerance in the United States and are aware of the 28-day preslaughter interval.

FUTURE TRENDS

Farm practices reflect changes in political and economic pressures. Federal farm programs, such as recent changes in acreage allotments and projected yields for cotton, will undoubtedly cause re-examination of insect control practices. Optimum farm operation may not emphasize maximum yields, and both crop choice and production practices will be re-evaluated for their net return to the producer. This could lead to a reduction in the amounts of pesticides and other economic inputs used for the production of certain crops.

There is a continuing need for insect control in crop and livestock production, but only a limited number of new pesticides are in view. Older materials, such as toxaphene, as long as they are environmentally acceptable, will continue to be used where they are still effective. It should be recognized, however, that toxaphene has probably reached its maturity and while a rather stable volume is used, no great expansion in its use can reasonably be foreseen.

The use of toxaphene on cotton (Table 1) far exceeds that on any other agricultural commodity. Thus, any changes in future cotton insect pest control strategies may greatly affect the amounts of toxaphene used in the United States. Toxaphene is rarely used alone for the control of the insect pests of cotton. Historically, the greatest use of toxaphene on cotton has been in mixtures with DDT. The toxaphene-DDT mixture was synergistic to the chlorinated hydrocarbon insecticide-resistant strains of boll weevils, thrips and cotton fleahoppers which developed during the 1950's.

The combination of toxaphene with DDT provided a pesticide that was many times more toxic to the above pests than either component when used alone. The toxaphene-DDT mixture, marketed in a formulation containing two parts of toxaphene to one of DDT, provided very effective and economical control of the three above pests as well as the bollworm and tobacco budworm. The mixture also was very safe for applicators and farm workers to handle. Because of these reasons, toxaphene-DDT has been, and probably continues to be, one of the most widely used pesticides on cotton.

If the use of DDT on cotton should be prohibited, this action undoubtedly would have an effect on amounts of toxaphene used on the crop. In states where DDT has been banned (Arizona and California), or where the toxaphene-DDT mixture has lost its utility because of the continued development of insecticide-resistant insect pests (Texas), the DDT component of the mixture has been replaced with methyl parathion. However, the toxaphene-methyl parathion mixture is not synergistic against resistant insects, but simply additive. That is, each component provides toxicity to a given pest in direct proportion to the toxicity of the component when used alone.

In toxaphene-methyl parathion mixtures, methyl parathion is by far the most toxic component to insects. However, the addition of toxaphene to methyl parathion provides certain advantages in that the resultant pesticide is measurably more toxic to pest species than either insecticide alone. The toxaphene-methyl parathion mixture also is more persistent than methyl parathion; thus, the mixture may be applied with less frequency than methyl parathion alone. In comparison to toxaphene-DDT, the toxaphene-methyl parathion mixture poses a much greater acute hazard to applicators and farm workers. This mixture also is much more toxic to certain beneficial species of insect parasites and predators than toxaphene-DDT. The amounts of toxaphene applied in mixtures with methyl parathion oftentimes is less than in the traditional toxaphene-DDT mixtures.

Future trends for the use of toxaphene on other crops and livestock is not expected to change greatly. Toxaphene presently has considerable utility in the production of small grains, pasture and hay crops, soybeans, vegetables and livestock. It also has some use for the control of certain insect pests of corn, grain sorghum and other feed and food crops. Toxaphene has a particular advantage (as will be discussed in detail later in this report) in that it may be used on seed alfalfa, clover and certain vegetable crops without causing great damage to bee-pollinators.

CHEMISTRY AND COMPOSITION

Chemical structure and production of toxaphene. Toxaphene is defined as chlorinated camphene (67-69% chlorine) and has the empirical formula $C_{10}H_{10}Cl_8$ with a molecular weight of 414.

The commercial production of toxaphene (U. S. Patents 2,565,471 and 2,657,164, Hercules) consists of the reaction between camphene and chlorine activated by ultraviolet irradiation and certain catalysts to yield the final product of chlorinated camphene with a chlorine content of 67-69%. The final product is a relatively stable material with a mild terpene odor and is a mixture of related compounds and isomers.

Physical Properties

Physical form: Amber, waxy solid.

Melting point: 70°-90°C.

Solubility: High solubility in most organic solvents, but greater in aromatic solvents; water solubility is about 0.5 ppm.

Vapor pressure: 0.2-0.4mm/25°; 3-4mm/90°C.

Product Specifications

Total organic chlorine, % by weight	67.0-69.0
Acidity, % by weight as HCl	0.05% max.
Drop softening point, °C	70 min.
Infrared absorptivity at 7.2u	0.0177 max.
Specific gravity at 100°C/15.6°C	1.600 minimum

Typical Properties (Not Specifications)

Specific gravity at 100°C/15.6°C	1.63 (average)
Specific gravity change per °C	0.0012
Pounds per gallon at 75°C	13.8
Viscosity, centipoises at 110°C	89
120°C	57
130°C	39.1
Specific heat, cal/g/°C at 41°C	0.258
95°C	0.260

Uniformity of toxaphene production. Toxaphene produced by Hercules is regularly bioassayed and subjected to chemical and physical tests lot-by-lot during the manufacturing process. The housefly is a convenient test organism, although bioassay with other insects such as *plum curculio* and Southern armyworm is also recommended to agencies seeking standards of identity appropriate for specifying, purchasing or evaluating toxaphene insecticides.

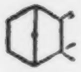
Recently, a series of nine samples from retained toxaphene production manufactured by Hercules in the interval 1949-1970 was bioassayed against female houseflies by the topical method. The laboratory toxaphene standard sample was used for comparison. Infrared absorption spectra and electron capture gas chromatograms were also prepared. Results show that the toxaphene regularly produced by Hercules during the past 23 years is quite uniform in its properties.

Composition of toxaphene. A large number of chlorinated compounds are present in toxaphene. A typical gas chromatogram suggests that 30 or 40 principal constituents may exist. The chlorine content in the commercial product is limited to 67-69% since insecticidal activity peaks sharply in that band.

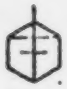
Control of camphene feedstock quality and process variables is important in achieving a material of uniform properties. Listed previously are product specifications established by Hercules for toxaphene produced by that manufacturer. The specification item of infrared absorptivity at 7.2μ helps distinguish toxaphene from other chlorinated terpene products such as Strobane.

Toxaphene is prepared by the chlorination of the bicyclic terpene camphene to contain 67-69% chlorine. The empirical formula for this material is $C_{10}H_{10}Cl_8$. Chlorination-grade camphene is prepared by the isomerization of α-pinene, a product derived from the Southern pine tree. Some tricyclene may accompany the camphene, but less than 5% other terpenes are present.

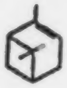
Structures of some of these perpenes are as follows:



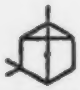
camphene



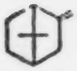
(II) tricyclene



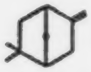
(III) α-pinene



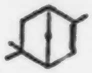
(IV) cyclofenchene



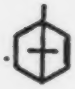
α-fenchene



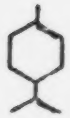
(VI) β-fenchene



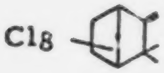
(VII) γ-fenchene



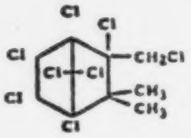
(VIII) bornylene



dipentene



(X) toxaphene



(XI) toxaphene according to Messing (3)

The structure X is commonly used to depict the structure of toxaphene. The only published chemical structure that is more detailed than X is that suggested by Messing (3), who proposed structure XI, though apparently with qualifications (1, 2).

Due to the complexity of the chemical reactions in the synthesis of toxaphene, a large number of components is present in the product. Separation of these components by a variety of means has been attempted. A description of some of these results follows.

Partition chromatography. A system of heptane on carbon and 90% aqueous methanol was most useful in separating toxaphene components. However, sharply defined peaks were not obtained. Melting points ranged from 15°C to as high as 210°C, but none were sharp. Only slight differences in infrared absorption spectra were observed. Insecticidal activity of various fractions did not differ widely.

Fractional crystallization. A typical fractional crystallization system applied to toxaphene utilized isopropanol solvent and carried through 5 levels, combining mother liquors and crops to obtain additional fractionation. Five crops (3 crystalline and 2 non-crystalline) were obtained. Melting points varied widely, but insecticidal activity as measured by fly bioassay did not differ much. A summary of the results is shown in Table 1.

TABLE 1

Properties of Fractions from Fractional Crystallization of Toxaphene

		%Kill (Flies -- Bell Jar)			
		0.1% Conc.		0.05% Conc.	
Sample	Melting Range	AV.	S. D.(b)	AV.	S. D.(b)
Toxaphene	-	56(9)(a)	11.3	33(8)(a)	16.1
22	234-239°C	70(9)(a)	5.4	39(8)(a)	8.1
24	208-210°C	80(9)(a)	8.8	40(8)(a)	11.8
26	184-187°C	78(9)(a)	9.3	40(8)(a)	11.4
28	Noncrystalline semisolid	44(9)(a)	8.1	29(8)(a)	13.4
30	Viscous liquid	40(9)(a)	8.3	22(8)(a)	7.5

(a) Numbers in parentheses are numbers of determinations.

(b) S.D. = standard deviation of test results.

Craig liquid-liquid separation. A 100-stage Craig liquid-liquid extractor was used with solvent pairs that included isooctane-acetonitrile isooctane-methyl cellosolve and isooctane-dimethyl formamide. The lack of sharp peaks indicated isolation of individual components was not obtained, but the broad spread of the resolved sample and the uneven contour of the Craig profile do indicate some separation. The biological data for the indicated fractions are tabulated below. The system isooctane-acetonitrile concentrated about 10% of the sample in the most polar phase, and the material was relatively nontoxic to flies.

Fractions separated in the system isooctane-methyl cellosolve were tested individually. The results show material of lower toxicity to be at both ends of the most polar-least polar spectrum. The fractions between the extremes seem to approximate the toxicity of the middle fractions of the isooctane-acetonitrile system.

TABLE 2

Craig Countercurrent Fractionation of Toxaphene

	% of Original	% Fly Kill at Indicated Concentration Topical Application			
Fraction No.	Sample	0.6 mg	0.5 mg	0.4 mg	Solvent System
X9675-23-A	11.4	3	0	0	Isooctane-Acetonitrile
-B	33.8	41	22	0	
-C	37.8	100	100	79	
-D	9.9	75	54	19	
-E	7.2	35	3	0	
Toxaphene Standard		91	81	28	
X9675-31-A	Tubes 5, 10, 15	7	0	0	Isooctane-Methyl Cellosolve
-B	Tube 45	31	22	16	
-C	Tube 85	100	97	57	
-D	Tube 125	79	63	28	
-E	Tube 185	0	3	0	
Toxaphene Standard		91	57	29	

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METHODS OF ANALYSIS[1/]

Assay procedures (formulation analysis). The procedures for toxaphene assays were described in two recently published books (17, 34), and are based on the following technologies:

(1) Total chlorine method (metallic sodium reduction).

(2) Total chlorine method (sodium biphenyl reduction).

(3) Infrared spectrophotometry.

(4) Colorimetric spectrophotometry (diphenylamine-zinc chloride).

Total chlorine methods. In practice, an isopropyl alcohol solution of the toxaphene sample is treated with metallic sodium; or a benzene solution of the sample is reduced with sodium biphenyl reagent. The liberated chloride is then titrated by the nitrobenzene modification of the Volhard procedure. An alternate organic chlorine method for toxaphene-sulfur dusts involves the liberation of chloride by the Parr peroxide bomb method and the determination of chloride as above.

Infrared spectrophotometry. Toxaphene formulated as a dust, wettable powder or emulsifiable concentrate may be assayed by Clark's (9) infrared method, which may also be used to measure toxaphene and DDT simultaneously. Concentrations of each component are read from calibration curves prepared from CCl_4-solutions of known toxaphene/DDT content, by reading maximum and minimum absorbancies at 7.8μ and 6.0μ, respectively for toxaphene and 9.1μ and 5.8μ for DDT.

1/ Contributed in part by F. J. Carlin, Hercules Res. Center

Spectrophotometric method. Spectrophotometric methods may be used to assay toxaphene formulations. The procedure of Graupner and Dunn (20), which involves the development of a greenish-blue color by the fusion of toxaphene with diphenylamine in the presence of zinc chloride, has been applied to assay and residue analysis.

Two other methods were evaluated by Hercules. The colorimetric procedure developed by Nikolov and Donev (32) using alkali and pyridine to develop a reddish-brown color appears to be unsatisfactory because of poor precision and accuracy. However, a procedure developed by Hornstein (21) using thiourea and KOH to give a yellow color seems to be satisfactory for toxaphene assay.

Assays for cattle dips. Total chlorine and infrared spectrophotometric procedures were applied to the analysis of toxaphene in cattle dips. Infrared procedures are more specific for toxaphene.

F. P. Czech (13) developed a rapid infrared method for toxaphene in animal dips and sprays, which was based on the method by Clark (9). The USDA also published a "Testing Procedure for Emulsifiable Concentrates of Toxaphene," which presented a compilation of total chlorine and infrared procedures (41) applied to livestock dip analyses. In a series of publications, Czech presented a rapid vatside test for toxaphene and many chlorinated hydrocarbon insecticides (14, 16). The preferred method (16) involved "salting-out" the insecticide, extracting it into an organic solvent, removal of chlorine with sodium biphenyl reagent and coulometric titration of the chloride liberated. Using an automatic coulometric titrator improved the precision of the analysis.

Both the total-chloride and colorimetric spectrophotometric methods have been utilized for the analyses of toxaphene residues in agricultural crops and foods. However, these methods suffer from non-specificity (total chloride) and lack of sensitivity (total chloride and colorimetric). Infrared spectrophotometry has never achieved the required sensitivity to become practicable for residue determinations.

Residue analyses for toxaphene. Until 1960, no analytical residue methods for pesticides involved gas chromatographic techniques (11). Thus, any pesticide residue data reported in the literature, at least until 1960 , but more probably until 1963, were obtained by conventional residue methods, e.g. spectrophotometry. This assumption must also be made for toxaphene. The two methods of choice for residue analyses of toxaphene until about 1963 were: total chlorine determination and colorimetric spectrophotometric method.

As stated before, the total chloride method suffers from non-specificity and the spectrophotometric from low sensitivity; both methods require rigorous cleanup due to possible interferences from plant or animal extractives.

Since about 1963, reported toxaphene residues in crops, foods, tissues and other natural samples were probably obtained by gas chromatography. Due to the heterogenous composition of toxaphene and related chlorinated camphene products, these reports must be carefully scrutinized. The inherent difficulty for toxaphene analysis is also shared by other chlorinated pesticides like Strobane and chlordane and will be discussed in greater detail below.

In 1966, Archer and Crosby (1) described a pre-treatment of samples suspected to contain toxaphene. This resulted in a gas-chromatographic elution pattern more suitable for qualitative and quantitative determination of toxaphene residues than the multi-peak pattern of untreated samples.

The treatment consisted of a partial dehydrohalogenation of toxaphene by KOH in ethanol resulting in three major peaks emerging sooner than DDE, the dehydrochlorinated product of DDT . This method was modified by Hercules chemists and forms the basis of the recommended method of toxaphene residue determinations.

Other ancillary techniques for residue determinations of toxaphene are paper- and thin-layer chromatography, but these suffer from the same diffuse patterns or multi-spots as the earlier gas chromatographic technique.

Clean-up procedures. Two techniques are widely used to clean up extracts for toxaphene residue analysis (35). Absorption chromatography on Florisil permits removal of plant pigments and some waxes; also, separation of toxaphene from a few chlorinated hydrocarbon insecticides and most thiophosphate materials is accomplished by elution of toxaphene with 6% (v/v) diethyl ether in hexane. Fats and oils are separated from toxaphene by contact with concentrated sulfuric-fuming sulfuric acid mixtures. A 1:1 mixture of the sulfuric acids is ground with Celite 545 and packed into a chromatographic column. A hexane solution of the fatty material is applied to the top of the column. Toxaphene is eluted with hexane, while the sulfonated fats and oils are retained on the column.

After nitration of extracts, DDT was removed as an interference in toxaphene residue analysis (18). Also treatment with concentrated sulfuric-fuming nitric acid mixtures did not alter the analytical characteristics of toxaphene (23).

Two procedures for eliminating polychlorinated biphenyl (PCB) interferences from chlorinated hydrocarbon insecticide residues were evaluated. In a procedure by Reynolds (35), PCB's, along with heptachlor, aldrin and DDE are eluted from Florisil with 200 ml of hexane, but lindane, heptachlor epoxide, dieldrin, DDD and p,p-DDT required 250 ml of 20% ethyl ether in hexane for complete elution.

The procedure by Armour and Burke (2) involved elution of PCB's from a silicic acid/Celite 545 column with 250 ml of hexane, while DDT and its analogs were eluted with 200 ml of a mixture of 1% acetonitrile + 19% hexane + 80% methylene chloride. Both procedures were applicable to toxaphene; however, Reynolds' procedure is preferred. Armour and Burke's procedure requires prior cleanup on a Florisil column, but Reynolds' procedure is cleanup and separation on a single column.

Measurement of toxaphene residues may be accomplished by spectrophotometric methods, total organic chlorine determinations, or chromatography.

Total Chlorine Methods

Schöniger Combustion

A procedure for the determination of toxaphene residues in animal fat and butterfat involves combustion of the sample followed by amperometric titration of the liberated chloride with silver nitrate (22). Sensitivity of the method was 5 mg of toxaphene. Another combustion procedure applicable to toxaphene was published by Lisk (28). The procedure involves combustion of the sample in a Schöniger flask and spectrophotometric determination of chloride by displacement of thiocyanate in the presence of ferric ion.

Zweig, et al (44) combined the Schöniger combustion method, following fuming sulfuric acid treatment and amperometric titration of the liberated Cl^- ions to achieve an overall sensitivity of 0.02 ppm toxaphene in whole milk. However, the "total organic chlorine" method is recommended for samples of a known history, e.g. milk from cows fed known quantities of toxaphene.

Active-Metal-Reaction Methods

Sodium reduction techniques are widely used for residue analysis of chlorinated hydrocarbons such as toxaphene. Phillips and DeBenedictis (33) modified the sodium-isopropanol reduction method as applied to the determination of chlorinated pesticides.

Liggett (27) and Chapman (8) used sodium biphenyl to determine organic chlorine. Menville et al. (29) and Koblitsky et al. (25), utilized sodium dispersions for the decomposition of organic chlorine. The latter method deals specifically with the detection of chlorinated pesticides in animal fat.

The techniques preferred by Hercules for the determination of total organic chlorine consist of a sodium-liquid ammonia decomposition method followed by an amperometric titration using coulometrically generated silver ions. The decomposition method is based on the work of Beckman *et al*. (3).

For quantitative measurement of the chloride resulting from any of the above-mentioned techniques the automatic chloride titrator is preferred, based on an instrument described by Cotlove (10) and sold commercially by American Instruments Company, Silver Springs, Maryland, or Buechler Instruments, Inc., 514 West 147th Street, New York 31, New York.

This instrument has a silver coulometer to generate the reagent and an amperometric end-point detecting system that automatically stop the titration after the end point is reached. The time needed to complete a titration is recorded on a built-in electric timer. This time is easily related to chloride content of the sample.

Lisk (28) prefers the spectrophotometric determination of chloride based on the displacement of thiocyanate from mercuric thiocyanate in the presence of ferric ion. The technique is suggested as an alternate detection procedure for laboratories not equipped with the Cotlove titrator.

Spectrophotometric Method

The spectrophotometric procedure (20, 17) is a moderately sensitive method for qualitative and quantitative analysis. The greatest shortcoming of the method is the need for exhaustive cleanup because small amounts of plant waxes develop colors and interfere with the detection of toxaphene. The method may be used as a confirmatory technique, however.

Klein and Link (24), in their studies on toxaphene residues on kale compared residue data obtained by the diphenylamine method with gas chromatography data. Agreement was good at residue levels about 10 ppm. Blank color formation was significantly reduced after treatment of the crop extracts with a concentrated sulfuric-fuming nitric acid mixture.

Paper Chromatography

Paper chromatography is used to detect and estimate chlorinated organic pesticide residues (30). The limit of detection is about 0.2 micrograms of toxaphene, but chromatograms result in streaks (38).

Thin-Layer Chromatography

Thin-layer chromatography (TLC) resembles paper chromatography as a technique, but provides the added advantages of greater speed, and frequently, higher sensitivities.

The preferred TLC procedure is similar to that of Schechter (37) and Moats (31). The TLC system employs layers of aluminum oxide and the chromogenic agent, silver nitrate, added to the absorbent when the

TLC plates are prepared. The plates are spotted and developed in the normal manner using hexane as the mobile phase. After solvent development, the plates are exposed to UV light to reveal toxaphene at the 0.5 microgram level.

Gas Chromatography

Review of Methods

It became apparent from the first work on gas chromatography that chlordane, Strobane and toxaphene resulted in at least seven peaks (12, 19) (See Fig. 1). Witt (43) tried to reduce these multi-peaks into a single peak using a 1 1/4-ft-long column instead of the conventional 6-ft length. Using microcoulometry, 0.5 μg of toxaphene could be detected at a retention time of less than 2 min (see Fig. 2).

This method was used to determine toxaphene levels in water, aquatic plants and fish from lakes treated with toxaphene (40). Apparent levels of toxaphene in untreated control samples ranged from an average of 0.38 ppb in water to 0.55 ppm in fish. However, using a short gas chromatography column decreases the resolution of toxaphene isomers and related compounds as well as other commonly occurring pesticides. Thus, absolute identification of single peaks is almost impossible.

To improve the method of identifying toxaphene residues by gas chromatography, Bevenue and Beckman (3) fingerprinted toxaphene by three major characteristic peaks on a 5% QF-1/Chromosorb-W column, eluting after DDT, thus differentiating between DDT and toxaphene. The detectibility of toxaphene with an electron-capture detection is claimed to be

2 ng under ideal conditions but more usually 5-7 ng. To stress the limitation of this method, these authors state,

> ".... the pesticide residue chemist has been placing increased reliance on gas chromatographic data for the identification of a pesticide residue. In the examination of a sample for toxaphene residue, such data are not reliable, either qualitatively or quantitatively. In particular, when state or other regulatory agencies may wish to examine a shipment of produce suspected of excess toxaphene residue, the use of gas chromatography data alone for the basis for legal actions is an invitation for criticism and rebuttal.
>
> "We believe the same thesis could be applied to the compounds chlordane and Strobane. Until it can be shown by some new, and presently unknown, technique that toxaphene can be unequivocally identified, the gas chromatographic procedure for the determination of toxaphene, alone or in combination with other pesticides, is at best highly questionable. Further investigation into the '3-peak' phenomena at the latter part of the gas chromatographic curve may possibly produce a definitive fingerprint." (See Fig. 3).

Gaul (19) has recommended the planimetry of the last four peaks as a quantitative measurement of toxaphene in the presence of DDT. If Kelthane is present, superimposing a toxaphene standard at about the same concentration as the unknown sample will correct the situation.

The last four peaks of a toxaphene chromatogram are not always observed, and samples containing toxaphene should be treated with concentrated sulfuric acid - fuming nitric acid (18). The acid treatment does not appreciably alter toxaphene and chlorinated camphene, but it effectively removes residues of DDT, aldrin, heptachlor, Kelthane, Perthane, Tedion, Telodrin and Trithion (23).

Archer and Crosby (1) measure chromatogram quantities of toxaphene in milk, fat, blood and alfalfa hay with a simple alkali treatment for cleanup, partial dehydrohalogenation, and electron capture gas chromatography on a column of 5% DC-710 silicone oil and 5% silicone oil and 5% SE-30 at 200°C. They used a single modified toxaphene peak eluting at 3.50 min for quantitative analysis and qualitative identification. This peak has a shorter retention time than the modified peaks of the DDT group (DDE and related compounds) commonly present in samples (see Fig. 4),

Recommended Procedure[2/]

The recommended method for the residue analysis of toxaphene involves a sulfuric acid-Celite 545 column cleanup followed by dehydrohalogenation and gas chromatography, which is a modification of the work of Archer and Crosby. The sulfuric acid column removes fats and oils, and the dehydrohalogenation gives a characteristic, reproducible pattern for dehydrohalogenated toxaphene.

2/ Carlin, F. J. Jr., Hercules Inc. (1970)

The sample to be analyzed is dissolved in a small amount of n-hexane and passed through a H_2SO_4-Celite column with 100 ml of redistilled n-hexane. The hexane is evaporated and the sample treated with ethanolic 25% KOH at 75-80° for 15 min. The reaction mixture is diluted with water and extracted with 0.5 ml n-hexane. Aliquots of the hexane layer are gas-chromatographed.

Gas chromatography is performed on a 9-ft. x 1/8 in. column, 1:1 mixture 5% SE-30, 5% DC-710 silicone oil on (100/120) Gas Chrom Q; column temperature 200-210°; electron capture detector. Column conditioning for 2 days at 250° is highly recommended. The area of major peak of dehydrohalogenated toxaphene eluting at about 4.5 min or the entire trace is measured by triangulation and used for quantitative analysis.

If additional cleanup of sample is needed, this can be done by Florisil chromatography, toxaphene eluting with the "6% ethyl ether in petroleum ether" fraction.

Thirty nanograms of toxaphene produced 80% of full-scale deflection with a 1 mv-recorder (1).

Recommended gas chromatographic conditions for unmodified toxaphene are the following: 5 ft. x 1/8 in. - glass column packed with 3.8% UCW-98 on Diataport S (80/100 mesh); column temperature 150°C; carrier gas (N_2) flow - 45 ml/min.

Discussion of Analytical Methods and Reported Data

The analysis of toxaphene by gas chromatography shows that due to the heterogeneity of the compound, a definite identification of toxaphene by distinct peaks or fingerprints is unsatisfactory. Chemical modifications by acid-treatment and/or dehydrohalogenation result in a distinct improvement of the elution pattern. Samples with a known spray history can be analyzed by most of the analytical methods described above including total chlorine, spectrophotometric and gas-liquid chromatography.

However, environmental samples of soil, water, air, fish and wildlife and human specimens, which have been analyzed for chlorinated pesticides by gas-liquid chromatography without prior chemical treatment cannot be unequivocally analyzed for toxaphene residues.

For example, Burke and Giuffrida (7) report the retention times, relative to aldrin, of the major peaks of toxaphene on 10% DC200 at 200^{o} and a carrier gas flow of 120 ml/min, to be:

2.34; 3.06; 3.61; 4.51 (Aldrin = 1.00)

Under the same conditions DDD has a relative retention time of 2.33 and p,p'-DDT, 3.03. Gaul (19) illustrates that methoxychlor has the same retention time as one of the major peaks of toxaphene (No value is given, but it is possibly the 4.51 min peak quoted by Burke and Giuffrida, 7).

An attempt, therefore, was made to evaluate reports of the presence or absence of toxaphene residues in natural samples of unknown spray history in order to make a judgment of the validity of the reported findings. Some of these reports are summarized in Table 1.

Table 1 does not give detailed summary of toxaphene residues found in crops, tissues or food, but rather illustrates the gas chromatographic technique used and the apparent success to analyze for toxaphene with a high degree of certainty. Of the 10 examples chosen on the basis of "toxaphene" in the title and published during the past 10 years, only one author, Archer (1) uses the chemical pre-treatment method. All other reports on toxaphene residues cited in Table 1 rely on the multipeak phenomenon of toxaphene and some authors (examples 3, 5, 8, 9, Table 1) actually state their inability to identify toxaphene due to the complexity of the GLC elution pattern.

Conclusions on Analytical Techniques and Evaluation of Residue Data

1. Any samples for the analyses of toxaphene should use from hereon the recommended method involving acid clean-up and partial dehydrochlorination prior to GLC.
2. Past reports on the monitoring of toxaphene should be scrutinized for statements of sensitivity of method and any special pre-treatment of samples prior to analysis. In future work on toxaphene, explicit statements concerning lower limits of detection based on fortified samples must be included.
3. While the modified GLC method for toxaphene is superior to previously reported general GLC, it is subject to additional improvement for specificity and sensitivity. Research along these lines is encouraged.

4. It would be highly useful to re-examine where possible, retained samples, as for example environmental samples, by the improved GLC clean-up procedure for toxaphene and to verify previously reported results.
5. Decline and feeding studies with a known treatment history must be considered to be reliable by whatever recognized analytical techniques were employed, including total-chlorine, spectrophotometric or "GLC-no treatment" methods.

TABLE 1

Summary of Reported Toxaphene Residues by GLC

Sample	Toxaphene residue (ppm)	Method of analysis	Lit. Reference
1. Ladino clover seeds			
1 lb/A DDT; 2 lbs/A toxaphene; May 25, 1965 1.5 lb/A DDT; 3 lbs/A toxaphene; 1.5 lb/A Aramite; July 5, 1965 Analyzed in May 1966	65.7	GLC - dehydrochlorination	(1)
Ladino clover seeds (unknown history)	16.0	do.	do.
Ladino clover seeds (unknown history)	6.3	do.	do.
2. Kale, 2 lbs/A toxaphene			
Days after application: 0	155.0	GLC-EC (no treatment) (Area of 3 major peaks)	(24)
3	44.3		
7	16.9		
14	1.4		
21	0.3		
28	0		
3. 101 commercial animal feeds containing 87 dairy feeds or supplement; 15 samples were positive	0.06-0.53$^{(a)}$	GLC-EC (no treatment)	(30)
4. Oysters, 133 samples 6 positive samples	med. 0.08	GLC-EC (no treatment)	(6)
5. Milk	---$^{(b)}$	GLC-EC (no treatment)	
6. Drinking water	None detected$^{(c)}$	GLC-EC (no treatment)	(36)
7. Air samples	2520 ng/m$^{(c)}$	GLC-MC	(39)

TABLE 1 (Cont'd)

	Sample	Toxaphene residue (ppm)	Method of analysis	Lit. Reference
8.	Apples, broccoli, cabbage, grapes, lettuce, tea, carrots, potatoes, cabbage	---[d]	GLC-EC (no treatment)	
9.	Fish tissue	---[e]	a. GLC-EC b. Clean-up, TLC IR	(5)
10.	Cows fed 15 mg/kg of toxaphene for two weeks			
	Whole milk	25.3 (av.)	GLC-EC (no treatment; measure 3 major peaks)	(26)
	butter	27.9		
	cheddar cheese	28.1		
	dry whole milk	24.9		

(a) Authors state that presence of toxaphene is somewhat uncertain and the presence of toxaphene must be inferred from the general shape of the chromatogram; the larger values are probably more reliable.

(b) Authors state that toxaphene and Strobane could not be calculated.

(c) Less than 2.5 ppb.

(d) Authors state that chlordane and toxaphene could not be detected because of their multi-component nature.

(e) Authors state that the infrared spectrum of toxaphene was not clear for positive identification at a level of 50 μg or 2.5 ppm.

MC = microcoulometry

EC = electron capture

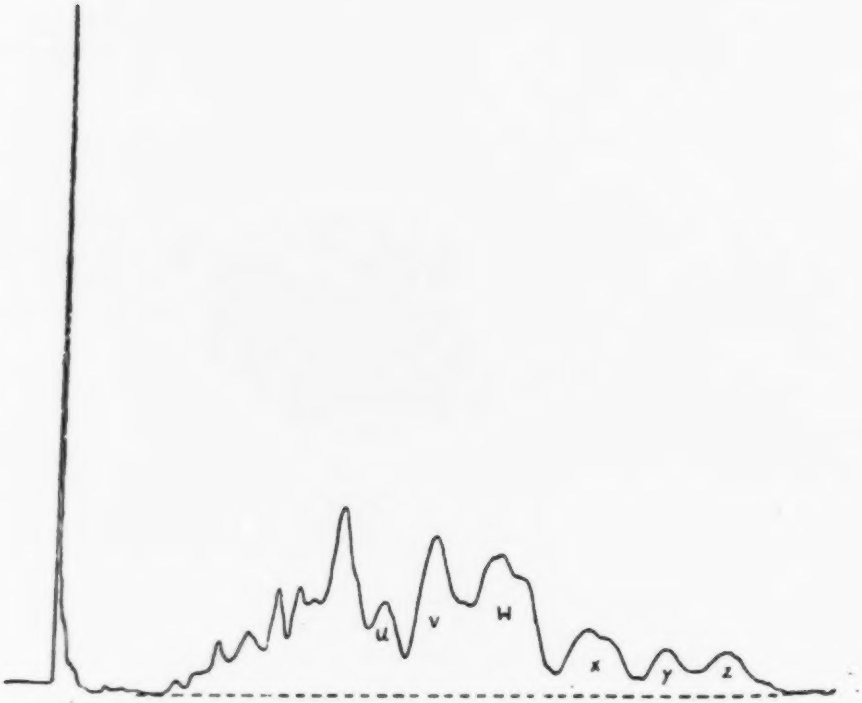

FIG. 1 Typical gas chromatogram of toxaphene (10% DC-200 on Anakrom ABS (90/100; Tritium electron capture detector; Column temp. 200°C; 125 ml/min nitrogen carrier gas) (19).

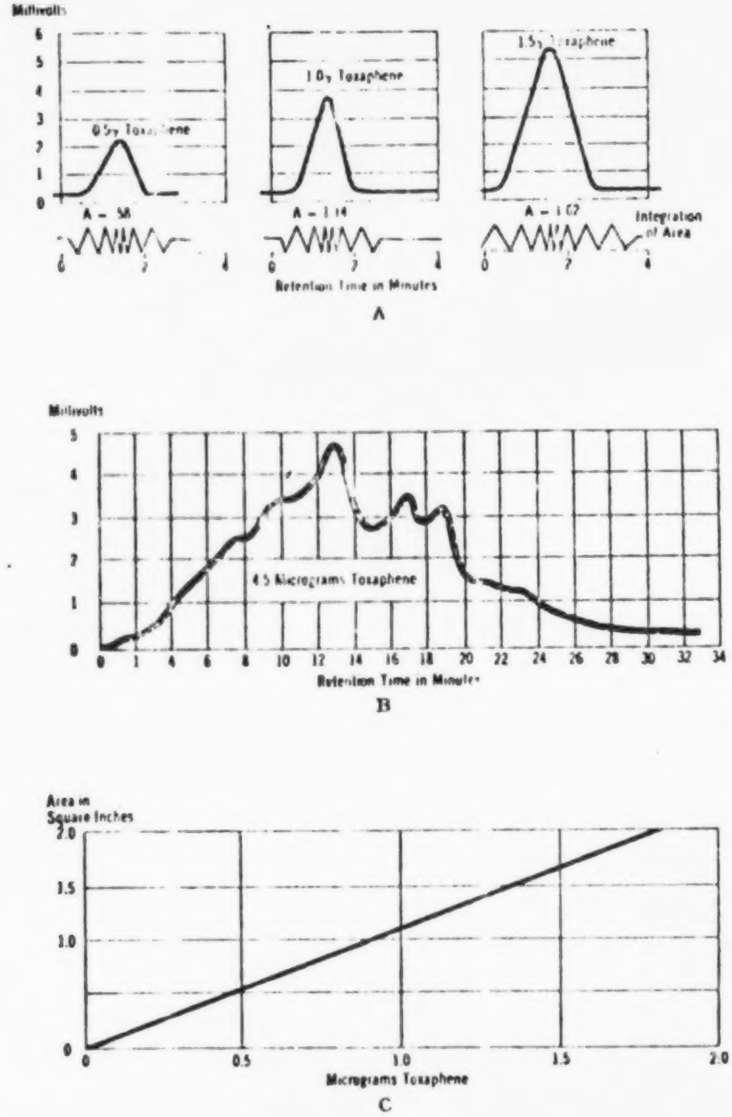

FIG. 2 Toxaphene analysis by gas-liquid chromatography and microcoulmometry (43).

A, 1 1/2-foot column; B, 6-foot column; C, standard curve of toxaphene.

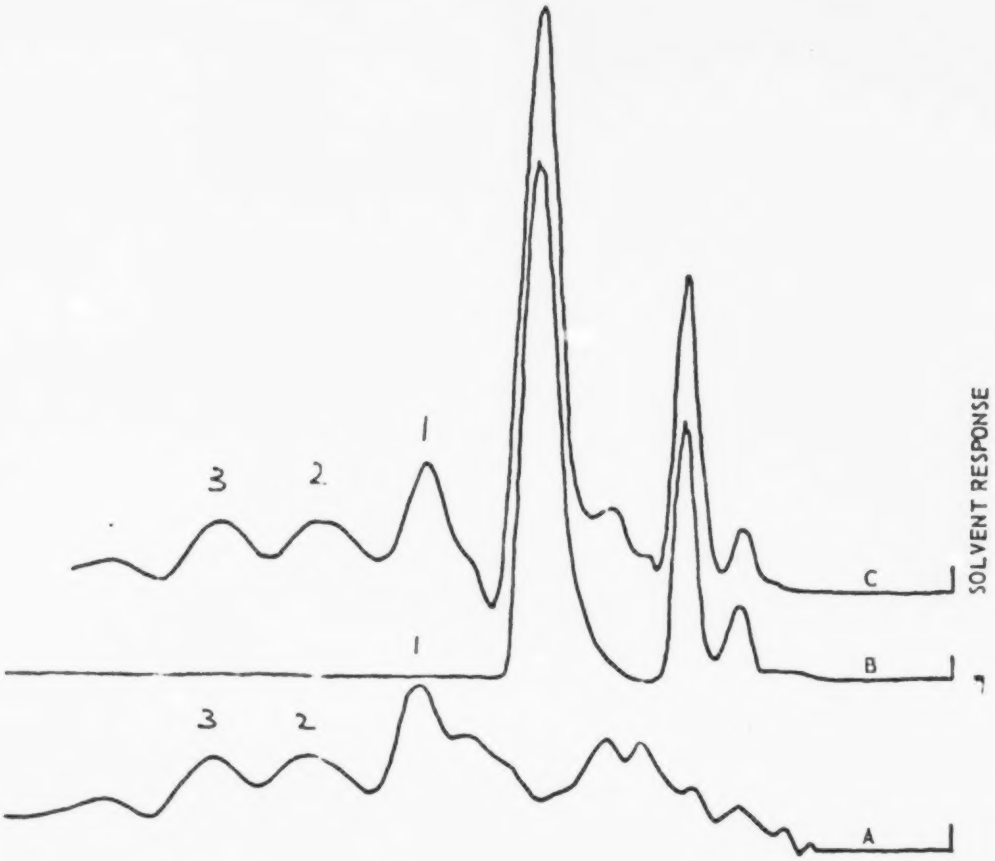

FIG. 3 Electron capture detector responses to: A-7 ng toxaphene, B-2.8 ng DDT, C-7 ng, toxaphene + 3.5 ng DDT (3).

COLUMN: 4' x 1/4" with 5% QF - 1 on Chromosorb - W.

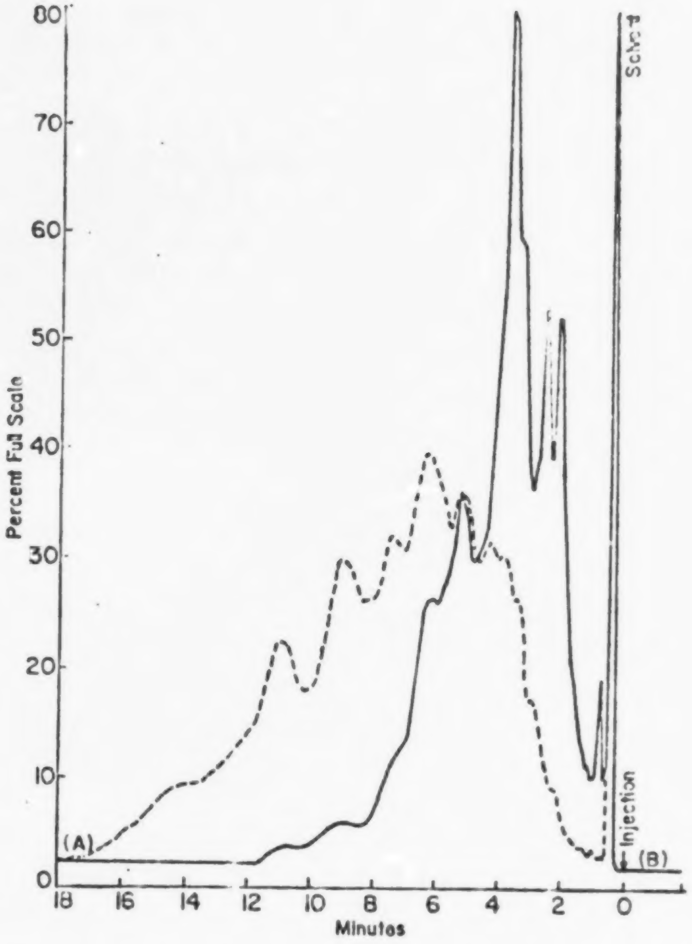

FIG. 4 Gas chromatogram from 30 ng. of toxaphene (A) before alkali treatment and after alkali treatment (B).

GLC conditions: 9' x 1/8" S.S. column mixed packing: 5% DC 710 and 5% SE-30 on chromosorb W (HMDS-treated) 12" section packed with CaC (20/30). Col. temp. 200° Nitrogen gas flow: 40-60 ml/min (1).

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FATE AND IMPLICATION IN THE ENVIRONMENT

EFFECTS ON FISH AND WILDLIFE

Toxicity and Pharmacological Actions

Acutely toxic doses of toxaphene administered to birds produced symptoms similar to those observed with other chlorinated hydrocarbon insecticides. The symptoms consisted of ataxia, goose-stepping ataxia, circling, low or high carriage, ptosis of eyelid, tremors, phonation, tenesmus, hyperthermia, wing-beat convulsions or opisthotonos. In some species of birds, symptoms were observed within 20 min; however, mortality usually took 2 to 14 days (22).

Acute toxicity of toxaphene was measured in several species of mammals, amphibia, birds, fish and invertebrates (21). In general, toxaphene exhibited a higher acute toxicity to fish and wildlife than DDT (Table 1).

Some feeding studies on quail and pheasant indicated an adverse effect on reproduction. Additional studies were conducted at the Patuxent Wildlife Research Center, Laurel, Maryland. However, tabulated results are not available.

Persistence of Toxaphene

The length of time that a pesticide will persist varies and depends on diverse factors such as temperature, rainfall, absorption, pH, microbiological populations and exposure to UV. Although toxaphene apparently dissipates rapidly from crops in a few days, in soil this may vary from several months to more than 10 years; and variations up to 9 years were seen in lakes and ponds (10, 11, 12, 17, 21).

Residues

Analyses for toxaphene are limited, probably due to lengthy and time-consuming procedures involved. In fish monitoring studies conducted by the Bureau of Sport Fisheries and Wildlife (8), toxaphene was found at low levels (0.01-1.25 ppm) in fish taken in Maine, New Jersey, Pennsylvania, South Carolina, Louisiana, Arkansas, Arizona and Utah.

In soil monitoring studies conducted by the U.S.D.A. (20), toxaphene was also observed in some samples. However, analyses for toxaphene were conducted only on a small number of the collected samples. Toxaphene analyses in birds, eggs, fish and reptiles are summarized in Table 2.

Analyses of catfish from fish farmers were conducted by the Bureau of Sport Fisheries and Wildlife at the Fish Pesticide Research Laboratory, Columbia, Missouri. Toxaphene residues in catfish fillet ranged from 0.3 ppm to 8.0 ppm; in mature channel catfish fat, 6-60 ppm (Av. 30); and in ovaries, N.D. to 3 ppm (Av. 1.8) (7).

Summary and Comments

After some initial testing, the Bureau of Sport Fisheries and Wildlife concluded that toxaphene should not be used as a pisciscide. It is toxic to fish and wildlife and may persist for extended periods, sometimes preventing the re-stocking of waters for several years. In waters treated with toxaphene, aquatic plants, benthic invertebrates and fish accumulate toxaphene. Fish may accumulate in their tissues several parts per million of toxaphene as long as a year after the treated waters are no longer toxic.

Monitoring studies indicate that toxaphene may be adsorbed to soil particles and carried into rivers. The mortality of fish-eating birds at the Tule Lake and Lower Klamath Refuges was associated with residues of toxaphene and other chlorinated hydrocarbons. Some birds were found dead after toxaphene was used in some Nebraska lakes and in Montana to control grasshoppers.

Research indicates that toxaphene "half-life" in soil may vary from 3-10 years; and in water, up to 6 years. The data also indicate that toxaphene can undergo bio-magnification, although to a lesser degree than most hydrocarbon pesticides.

Analysis of fish and wildlife tissues for toxaphene residues is most difficult and time consuming. Few laboratories can or are willing to undertake the task of analyzing large numbers of samples for toxaphene. This probably accounts for the fact that quantitative data for toxaphene is meager. Data for monitoring studies is sometimes conflicting and inadequate.

Improved sensitive analytical procedures, capable of screening many compounds, are needed for analysis of fish and wildlife tissues. Information is also needed in respect to the effects of toxaphene on the reproduction of birds. These should be available soon at the Patuxent Wildlife Research Center.

Beyond the fact that toxaphene toxicity dissipates in the environment, and that lakes and ponds become habitable for fish after toxaphene treatment, we have no significant information concerning the metabolism or degradation of toxaphene in the environment -- neither physiological nor chemical data. The void created by this situation must be filled.

TABLE 1

Toxaphene Residues

Organism		Toxaphene
Common Name	Scientific Name	48 hr LC_{50}(a) (ppm)
Bullhead, black	Ictalurus melas	0.005
Carp	Cyprinus carpio	0.0053
Minnow, fathead	Pimephales promelas	0.019
Goldfish	Carassius auratus	0.014
Sunfish	Lepomis cyanellus	0.018
Bass, largemouth	Micropterus salmoides	0.0051
Perch, yellow	Perca flarescens	0.018
Catfish, channel	Ictalurus punctatus	0.0172
Minnow, sheepshead	Cyprenodon Variegatus	0.007
Spot	Leiostomus xanthurus	0.0032
Bluegill	Lepomis Macrochirus	0.014
Trout, rainbow	Salmo Gairdneri	0.014
Killifish, longnose	Fundulus similis	0.028 (24 hr)
Mullet, striped	Mugil cephalus	0.0032
Salmon, coho	Oncorhynchus kisutch	0.012
Salmon, chinook	Oncorhynchus tshawytscha	0.008 (96 hr)
Trout, brown	Salmo trutta	0.0084
Toad, Woodhouse's	Bufo woodhousi	0.29
Frog, northern chorus	Pseudacris triseriata	0.7
Oyster, eastern	Crassostrea virginica	0.02 (96 hr)
Shrimp, brown	Peneus aztecus	0.0027
Shrimp, pink	Peneus duorarum	0.0042
Shrimp, glass	Palemonetes kadiakensis	0.006
Shrimp, grass	Palemonetes pugio	0.0052
Shrimp, Korean	Palemonetes macrodactylus	0.037
Daphnia	Daphnia pulex	0.015
Flea, water	Daphnia serrulatus	0.01
Scud	Gammarus fasciatus	0.022
Scud	Gammarus lacustris	0.07
Stoneflies	Pteronareys californica (Newport)	0.007
Stoneflies	Clossonia sabulosa (Banks)	0.0032
Stoneflies	Pteronarcella Bodia (Hagen)	0.0056
Damselflies	Ischmura verticalis	0.086

(a) These values may vary with temperature, pH, water hardness or the pesticide formulation itself.

TABLE 1 (CONT.)

	Sex	Age	LD_{50}(mg/Kg)*
Mallard ducklings	--	7 days ± 1	30.8 (23.3-40.6)
Mallards	♀	3-5 mo.	70.7 (37.6-133)
Pheasants	♀	3 mo.	40.0
Bobwhite quail	♂	3 mo.	85.4 (59.2-123)
Sharp-tailed grouse	♂	1-4 yr.	10-20
Fulvous tree ducks	♂	3-6 mo.	99.0 (37.2-264)
Lesser sandhill cranes	♀	--	100-316
Domestic goats	♂	>5 yr.	>160
Mule deer	♂	16-17 mo.	139-240

*95% conf. lim.

TABLE 2

TOXAPHENE RESIDUES IN WILD BIRD TISSUES

Species	Tissues Analyzed(a)	No. of Analyses	Range or Average of Residues found in ppm	Reference
Grebe, Western	Fat	5 analyses	0.0-39.0 Av. 12.66	16
Aechmophorus occidentalis	WB	8 analyses	0.0-0.8 Av. 0.02	16
1960	Carcass	6 analyses	Av.0.3	14
1960	Fat	2 analyses	Av.31.5	14
Gull, Ring-Billed Larus delawarensis	Fat	1 analysis	4.8	16
Heron, Black-Crowned Night				
Nycticorax nycticorax	WB?	No. not given	Up to 5.0	15
	WB	3 analyses	0.0-15.0 Av. 5.0	16
1961	Carcass	1 analysis	15.0	14
	WB found dead	1/1	64.0	14
Heron, Great Blue	WB	1/1	10.0	2
Ardea herodias	WB	1/1	10.0	16
	Carcass	1/1	10.0	14
Killdeer	WB	2/2	6.0	14
Charadrius vociferus	WB found dead	1/1	9.6	9
Kingbird, Western Tyrannus verticalis	WB young	1/1	4.0	14
Lark, Horned	WB sacrificed	4/4	0.41-0.96 Av. 0.7	9
Eremophila alpestris	WB found dead	3/3	Tr., 2.5. 3.3	9
Meadowlark, Western	WB found dead	3/3	Tr.,Tr., 0.6	9
Sturnella neglecta	WB	2/2	13.0	14
	WB young	3/3	3.0	14

(a) WB-whole body; L-liver; K-kidney; H-heart; BM-breast muscle

TABLE 2 (CONT.)

TOXAPHENE RESIDUES IN WILD BIRD TISSUES

Species	Tissues Analyzed (a)	No. of Analyses	Range or Average of Residues found in ppm	Reference
Blackbird, Brewer's *Euphagus cyanocephalus*	WB found dead	1/1	5.0	14
Coot, American *Fulica americana*	WB found dead	1/1	17.0	14
Cormorant, Double-Crested *Phalacrocorax auritus*	WB Carcass found dead	2/2 1/1	2.2-9.5 Ave. 5.8 9.5	16 14
Cowbird, Brown-Headed *Molothrus ater*	WB found dead	1/1	0.98	9
Dove, Mourning *Zenaidura macroura*	WB found dead	1/1	Tr.	9
Duck, Mallard Anas platyrhnchose	WB found dead	1/1	10.0	14
Duck Shoveler *Spatula clypeata*	WB found dead	1/1	12.0	14
Egret, Common *Casmerodius albus*	WB Carcass WB	1/1 3 analyses 4 analyses	17.0 Av. 9.2 0.0-17.0 Av. 6.92	2 14 16
Grebe, Eared *Podiceps caspicus*	WB	5 samples	0.0-4.0 Av. 1.9	16

(a) WB Whole body; L-liver; K-kidney; H-heart; BM-breast muscle

TABLE 2 (CONT.)

TOXAPHENE RESIDUES IN WILD BIRD TISSUES

Species		Tissues Analyzed (a)	No. of Analyses	Range or Average of Residues found in ppm	Reference
Pelican, White		L }-1 bird	1/1	8.0	2
Pelecanus erythrorhynchos		K)		13.0	2
		L }-1 bird	1/1	9.0	2
		K)		14.0	2
		1/2 bird (		4.0	2
		L 1 bird -	1/1	7.0	2
		K (			2
		H,L,K,BM	49 analyses	0.0-82.0 Av. 3.6	16
		L	3 analyses	7.0-9.0 Av. 8.0	16
		K	3 analyses	4.0-14.0 Av. 10.33	16
					13
	1960	Carcass	1 analysis	4.0	13
	1960	L	3 analyses	8.0	13
	1960	K	3 analyses	10.3	13
	1961	H,L,K,BM	12 analyses	7.6	13
Phalarope, Wilson's *Steganopus tricolor*		WB found dead	4/4	41.0	14
Sandpiper Sp. not given		WB found dead	1/1	10.0	14
Shrike, Loggerhead *Lanius ludovicianus*		WB sacrificed	1/1	Tr.	9
Teal, Blue-Winged *Anas discors*		WB	3/3	7.0	14
Wren, House *Troglodytes aedon*		WB	2/2	41.0	14

(a) WB-whole body; L-liver; K-kidney; H-heart; BM-breast muscle

TABLE 2 (CONT.)

TOXAPHENE RESIDUES IN WILD BIRD TISSUES

Species	Tissues Analyzed	No. of Analyses	Range or Average of Residues found in ppm	Reference
Pelican, White *Pelecanus erythrorhynchos*	H,L,K,M	Not given	82.0	22
Lark, Horned *Eremophila alpestris*	WB?	4 shot	0.7	22
	WB?	7 found dead	Tr. 9.6	22
Shrike *Lanius ludovicianus*	WB?	1 shot	0.7	22
Blackbird, Red-Winged *Agelaius phoeniceus*	Fat, B, K, L, H) Gizzard, m)	Not given	Tr. in all tissues	3

TABLE 2 (CONT.)

TOXAPHENE RESIDUES IN WILD BIRD TISSUES

Species	Eggs Analyzed	No. of Analyses	Range or Average of Residue found in ppm	Reference
Cormorant, Double-Crested *Phalacrocorax auritus*	Yolk	2 analyses	10.0	16
Duck, Gadwall *Anas strepera*	Yolk	5 analyses	Av. 0.04	16
Gull, Ring-Billed *Larus delawarensis*	Yolk	1 analysis	0.2	16
Pelican, White *Pelecanus erythrorhynchos*	Egg	22 analyses	0.0-6.7 A. 0.39	16
Tern, Forster's *Sterna forsteri*	Yolk	1 analysis	15.5	16

TABLE 2 (CONT.)

TOXAPHENE RESIDUES IN FISH AND REPTILES

Species	Tissues Analyzed	No. of Analyses	Range or Average of Residues found in ppm	Reference
Bass, Largemouth	Flesh	13 analyses	0.0-0.3 Av. 0.05	16
Micropterus salmoides	Viscera	8 analyses	0.2-2.0 Av. 1.13	
Bluegill	WB?	22 analyses	0.0-2.06AV. 0.48	4
Lepomis macrochirus				
Bullhead, Black	WB	89 analyses	0.37-15-2	12
Ictalurus melas				
Bullhead, Brown	Flesh	3 analyses	0.0-0.19 Av. 0.6	16
Ictalurus nebulosus				
Carp	Flesh	1 analysis	0.1	16
Cyprinus carpio	Viscera	2 analyses	0.0-0.1 Av. 0.05	16
Catfish, Channel	WB?	27 analyses	0.0-6.6 Av. 2.23	4
Ictalurus punctatus	Fat	8 analyses	0.4	16
Crappie, Black	WB	3 analyses	0.0-0.1 Av. 0.03	16
Pomoxis nigromaculatus				
Chub, Tui	WB	29 analyses	0.0-8.0 Av. 1.09	16
Siphateles bicolor				
Fish				
Sp. not given	WB	Not given	Tr.8.0	14
Pumpkinseed	WB	1 analysis	0.04	16
Lepomis gibbosus				
Fish	L	30 analyses	0.1-10.9 (8 samples)	19

TABLE 2 (CONT.)

TOXAPHENE RESIDUES IN FISH AND REPITLES

Species		Tissues Analyzed	No. of Analyses	Range or Average of Residues found in ppm	Reference
Salmon, Atlantic Salmo salar	(1962	Tissue extract	2 analyses	2.6-2.9 Av. 2.75	21
	(1963)	Tissue extract	2 analyses	1.11-5.5 Av. 3.24	21
	(1964)	Tissue extract	2 analyses	1.5-2.1 Av. 1.8	21
Shad, Gizzard *Dorsoma cepedianum*		Whole body?	17 analyses	0.0-4.75 Av. 1.49	4
Spot		Juvenile (No mortality but thickened gill lamallae at 0.1 and 0.01 ppb)			1
Leiostomus xanthrus		Juvenile (50% mortality within 6 days at 0.5 ppb)			1
Trout, Brown *Salmo trutta*		Tissue extract	5+ analyses	8.3-24.8 Av. 12.46	21
Trout, Rainbow *Salmo gairdneri*		Whole Body	37 analyses	0.43-5.4	1
	(1962)	Tissue extract	6 or more analyses	1.2-12.0 Av. 5.7	21
	(1963)	Tissue extract	6 or more analyses	2.75-13.7 Av. 7.72	21
	(1964)	Tissue extract	6 or more analyses	3.2-3.8 Av. 3.5	21
		Whole body	5/5	o.13,0.28,0.43,0.98,1.3	5
		Flesh	19 analyses	0.0-2.57 Av. 0.22	16
Turtle, Softshell *Trionyx spinifer*		Viscera	1 analysis	1.0	16

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FATE AND MOVEMENT OF TOXAPHENE IN TERRESTRIAL AND AQUATIC SYSTEMS

Persistence in soil. The fate and movement of a pesticide in and from the soil are influenced by the following broadly categorized factors: (a) the pesticide characteristics; (b) edaphic considerations; (c) climate; (d) topography; and (e) land use and management. Any of these factors that tend to promote the pesticide's persistence will tend to increase its potential for environmental dispersion.

Pesticide movement through or across soil is facilitated by the movement of water. Overland flow is generally more important in pesticide transport than passage through soil. Two processes are involved: (a) pesticide movement while dissolved in water and (b) pesticide movement while dissolved in water. Sodium humate, a natural organic compound found in water, can increase the water solubility of DDT by a factor of 20 (32). Thus, the solution and movement of other organic pesticides may be facilitated by a variety of dissolved or emulsified organic substances found in water. The water solubility of toxaphene is variously reported as 0.4 mg/1 and 3 mg/1. (10)

Bailey and White (2) stated that the principal means of pesticide transport within soils are: (a) diffusion in the airspaces of soil (b) diffusion in soil water; (c) downward flowing water; and (d) upward moving water.

Movement by diffusion through the soil and air spaces is important with pesticides having high vapor pressure such as soil fumigants. This process plays a dominant role in the eventual loss of pesticides from the soil by volatilization. Percolation is the principal means of movement of relatively non-volatile pesticides; diffusion in soil water is important in transport over very short distances. Upward movement may occur in irrigated areas where high evapo transpiration ratios are prevalent.

Therefore, the total amount of rainfall or irrigation water received, intensity (water flux), and frequency of received water all appear to affect pesticide movement in soils. These also influence overland transport and facilitate the entrance of pesticides into solution.

Most literature on toxaphene persistence in soil is disappointing in quality and quantity, especially that predating the era of general availability of gas-liquid chromatography. During this time, analytical results were based on nonspecific methods. Some studies at grossly exaggerated application rates or other abnormal conditions, are useful for specific purposes, but may be misleading in calculating the half-life of toxaphene. Abnormally high concentrations in the soil may overcome the ability of soil microorganisms to detoxify the compound. There is little information specifically related to toxaphene degradation by soil microbiota.

Mulla (23) applied toxaphene at the rate of 17.2 lb/acre to well prepared irrigated soil in California to evaluate Hippelates control methods. The toxaphene was disked into the soil. One month after application, effective control was 77%. The percentage of control remaining slightly over 2 years later varied from 13 to 19%. Shaw and

Riviello (26), in laboratory and small scale field tests with toxaphene applied topically to the soil at 50 lb/acre, found that effectiveness in killing Mexican fruit fly larvae declined to zero after 373 days. Thus persistence on the soil surface may be much less than when incorporated into the soil.

Bradley, et al., (6) working on small plots of Norfolk loamy sand and Goldsboro sandy loam soil in North Carolina on which cotton grew, applied toxaphene as foliar sprays of aqueous emulsion at approximately weekly intervals from early June until September 1969. The accumulated application was 23.9 lb/acre. Respectively, 10 and 5% remained in the soil in September of 1969; 4% was found the following March. Less than one percent was accounted for in water and sediment runoff.

Stevens, et al., (28) conducted studies nationwide at 51 locations in 1965-1967 to determine pesticide levels in soils. Samples were collected from 17 areas in which pesticides are used regularly, 16 areas with a record of at least one pesticide application and in 18 areas with no history of pesticide use. The only evidence of pesticide build-up was in some orchards that had been treated repeatedly with DDT over a number of years.

The data in Table 1 show that residues of toxaphene from crop applications over periods ranging from 1-14 years are present at only small fractions of the amount applied. In areas of regular pesticide use, 60% of the vegetable and/or cotton-growing fields sampled contained toxaphene/Strobane (0.66-9.38 mg/kg). Only one orchard (3%) and 12% of small grain and root crop-growing areas were positive. None was found in limited use and no use areas. In Montana toxaphene was applied at 1.5 lb/acre in diesel oil to range land and 44% could be accounted for in the soil after one day; only 3% remained after 84 days (8).

Nash and Woolson (24) determined the vertical distribution of toxaphene in Congaree sandy loam soil of Maryland that had received accumulated applications of 65 or 130 lb/acre during 1951-1953. Between 85 and 90% of the toxaphene remaining 13 years after the last application was found in the upper 23 cm, which corresponds to the cultivated zone. The quantity in the surface 7.6 cm was less than the mean quantity between 7.6 cm and 23 cm depths.

Volatility and photodecomposition may play an important part in dissipation of chlorinated insecticides in the surface layers. Thomas (30) working in natural watersheds in Texas studied the potential for insecticide vertical movement through soil to a depth of 5 ft. Very little toxaphene occurred below a depth of 1 ft. About 20% of the toxaphene applied in the preceding 10 years could be accounted for in the soil profile.

Formulation also apparently can influence the persistence of toxaphene. The United States Forest Service at Gulfport, Mississippi (27) is continuing studies of insecticides in soil to prevent termite damage. Toxaphene in No. 2 fuel oil applied to the soil surface at 1/2 pint/sq ft (0.4 lb toxaphene or 17,000 lbs/acre) was 100% effective for 16 years and 90% effective for 22 years. Soil depth penetration was estimated at 6 in.

However, when applied at the same rate as an emulsion (1/2 pt containing 0.4 lb toxaphene), only 80% effectiveness remained at the end of one year and 50% at 3 years.

The references indicate that toxaphene is a long-lived, but by no means "immortal" insecticide. Residues in and on the soil may be detected for several months to several years, but there is no evidence that build-up has occurred in the soil in areas of regular usage. Major losses occur from the soil surface by processes suggested but not well documented. These include microbial decomposition, photodecomposition and/or volatilization.

Incorporation of toxaphene into soil tends to prolong persistence. This insecticide is not normally used to control soil insects, but residues remaining on the soil from foliar applications may be turned under by cultivation and plowing. Downward migration through the soil does not normally occur to any significant degree. The formulation in which toxaphene is applied may also influence persistence. Studies are needed to clarify the fundamental mechanisms that control persistence and loss of toxaphene from the soil.

Note on the Half-Life of Toxaphene in Sandy Clay Soil

A recent report (14) gives half-life figures on a number of chlorinated pesticides, including toxaphene, in Holtville sandy clay. The application of toxaphene sprayed onto the soil surface and disked the same day into the upper 6 in. of the soil was as follows:

Year	lb/A active ingredient	
	toxaphene	DDT
1953	19.6	19.5
1954	20.0	20.0
1955	20.8	20.8
1956	20.8	23.2
1957	22.4	20.0

By regression analysis, the half-lives of toxaphene and DDT were 4 years.

Occurrence and movement in watercourses. The most intensive investigation in a single watershed of toxaphene occurrence in surface water is reported by Nicholson, *et al.*, (25) who studied a 400/sq mile cotton producing watershed in Alabama for 6 1/2 years. Water samples of 2,000 to 10,000 gal were processed through activated carbon adsorption units for recovery of insecticides. Analysis was by gas chromatography.

A peculiarity of the method was that water was extracted over periods 1 to 2 weeks thus averaging peak occurrences. The extended sampling period insured against missing toxaphene if its presence was discontinuous. The values were not absolute because of possible incomplete extraction from water and recovery from carbon. However, at least the indicated amounts were present. Efficiency may be about 50%. The sampling devices were operated almost continuously for the entire study period.

The cotton acreage varied annually from 12,700 to 16,500. Annual, toxaphene usage and recovery data are given in Table 2. The authors attribute the presence of toxaphene in Flint Creek primarily to surface runoff.

Significant findings of this study were: (a) low toxaphene concentrations (less than 1 μg/l) were recovered from Flint Creek and were associated with small cotton farm operations where ground equipment was primarily used; (b) the source of toxaphene was the watershed as a whole rather than a few favorably located fields; (c) occurrence in the samples was year around with largest recoveries in the summer application season; and (d) there were indications of a reduction in frequency of occurrence and in concentration in river water beginning about 6 mos. after the first of several seasons of much reduced toxaphene usage. The letter suggests the period required under Alabama conditions for land surface cleansing to begin.

Bradley, et al., (6) studies runoff from 180 sq. ft. instrumented plots in North Carolina on which cotton was grown and treated with toxaphene and DDT singly and combined. Less than one percent of the toxaphene occurred in the water and sediment running off these plots. Where DDT alone was used, 2.83% ran off, while 1.03% of DDT was found in runoff from those plots also treated with toxaphene. The rate of toxaphene application was about twice that of DDT.

These data do not imply that these percentages of toxaphene and DDT in runoff would reach lakes and streams. A large proportion of the transported insecticides were tied up on soil particles (96% of the DDT and 75% of the toxaphene) and is expected to deposit in the first low spot or settling area reached. Thus, the field location relative to a lake or watercourse is important.

Nearly 20% of all pesticides used in the United States is applied in California. Therefore, data from California have special significance. Irrigation farming is widely practiced and a peculiarity, in some areas, is the presence of underground tile drainage systems.

Bailey and Hannum (3) reported on the analysis of more than 630 samples taken in California of surface waters, agricultural drainage, sediments and aquatic organisms. Data for surface waters are given in Table 3. Although toxaphene was recovered at 14 of 20 sampling stations, concentration values were less than 1 µg/l. The concentrations found of DDT/DDD, DDE, heptachlor epoxide, lindane, dieldrin and BHC each were within the same range.

Somewhat more toxaphene was recovered in water from agricultural drains (Table 4). Pesticide concentrations were highest in areas affected by agricultural development and decrease in surface water in proportion to inflow dilution and uptake by sediments and aquatic organisms. The temporal distribution was related to agricultural drainage practices and to runoff from heavy rainfall.

Johnson, *et al.*, (18) studied pesticide concentrations in tile drainage and open drains in the San Joaquin Valley of California between 1963 and 1965. Toxaphene was detected in 13 of 66 analyses of tile drainage effluent in concentrations varying from 0.13 µg/l to 0.95 µg/l and averaging 0.53 µg/l. Water from surface drains that collected surface and subsurface water was positive for toxaphene 60 out of 61 samples. Concentrations varied from 0.10 µg/l to 7.90 µg/l and averaged 2.01 µg/l. The predominant residues found in surface water were DDT/DDD and toxaphene. The average concentration of toxaphene was higher than any other chlorinated hydrocarbon insecticide and it was found most frequently.

The annual reports of the San Joaquin District of the California Department of Water Resources (1963-1969) contain a wealth of data on toxaphene occurrence in Central Valley tile drainage effluent (Table 5) and in surface waste water drains (Table 6) from irrigated areas, in other Central Valley surface waters (Table 7), and in bay and ocean water (Table 8).

Twelve percent of 422 water samples from tile drainage systems contained toxaphene in concentrations ranging from 0.2 µg/l to 1.26 µg/l. Forty-eight percent of 447 agricultural surface water drains contained concentrations ranging from 0.04 µg/l to 7 µg/l. Due to the small degree of vertical movement through the soil demonstrated elsewhere for toxaphene, its recovery in underground tile drains in the concentrations indicated needs explanation.

There is a strong possibility of direct access of surface water to the drains under some conditions (5). Toxaphene was found in 12% of 712 other Central Valley surface waters in concentrations ranging from 0.02 µg/l to 0.93 µg/l, and in 47 bay and ocean water samples in concentrations of 0.03 µg/l to 0.60 µg/l.

Routine monitoring (7, 20, 22, 31) of waters of the United States has not indicated the presence of toxaphene. One reason may be that the amount required for detection in routine screening analyses is greater than that of most pesticides reported. Lichtenberg (21) states that the minimum toxaphene concentration required for recognition in his monitoring of 1 liter water samples is 1 µg/l, although lesser amounts may be determined in samples in which toxaphene presence is anticipated.

Toxaphene may be transported by water in solution or dissolved in organic constituents. It may also be absorbed on sediment that is suspended or deposited permanently or temporarily. Sometimes it is transported in or on the bodies of aquatic organisms.

The amount of toxaphene in sediment undoubtedly reflects the degree of usage as well as watershed soil management practices. Baily and Hannum (3) working in California reported toxaphene in sediment in much higher concentrations than they found in water (Tables 3, 4 and 9). Generally, sediments of smaller particle size had higher pesticide concentrations than did those of larger size.

Barthel, et al., (4) studied agricultural chemicals contained in stream bed materials of the Lower Mississippi River. Toxaphene/Strobane was found only in a 5-mile stretch in the vicinity of West Memphis, Arkansas. The concentrations varied from 100 to 600 μg/kg and were attributed to upstream agricultural usage.

Grzenda and Nicholson (9) studied cotton field soil, water, river bottom sediments, bottom fauna and fish at Flint Creek, Alabama, to determine the distribution of toxaphene, DDT and BHC among the biotic and abiotic components of a stream system. Soils from 33 cotton fields representing 206 acres were sampled. Data on insecticide residues in soils are given in Table 10 (See Table 2 for data on insecticide residues in water.)

No toxaphene was recovered from river bottom sediment, but DDT/DDE was found in 23 of 58 samples at 8 to 6400 μg/kg, and 6 contained traces of BHC. This was reflected in infrequent occurrence of toxaphene and BHC in bottom fauna, while DDT/DDE was found in all samples. All fish samples, however, contained toxaphene, DDT/DDE and BHC.

Nicholson et al., (25) showed the relative importance of sediment versus solution in the transport of toxaphene, DDT and BHC in Flint Creek, Alabama. Suspended sediment seemed less frequently involved in toxaphene and BHC transport than in DDT transport (Table 11). This suggests a lesser affinity for solid substrates of toxaphene in low water concentrations than that possessed by DDT, which is notoriously hydrophobic. Support for this contention comes from the fact that toxaphene was frequently recovered in clarified and treated municipal drinking water while DDT rarely was found.

Although toxaphene is not registered for use in fishery management, some of the experiences with its use for that purpose cast light on the fate of toxaphene in lake water. Various studies reported that toxaphene persistence was influenced by the concentration applied, sunlight, temperature, oxygen, alkalinity, hardness, turbidity, presence of bacteria, and pH, but no quantitative relationships were found between these factors and persistence. Previous conclusions were based on the time required for detoxification and restocking. Johnson, et al. (17) used gas chromatography to study the mechanisms of detoxification. Their results are given in Table 12.

This tudy shows that toxaphene may persist in a lake for several years after application for fishery management even though detoxification is rapid. All of the lakes were shallow and eutrophic. The authors point out that detoxification is accomplished, in part, by sorption reactions rather than degradation, but indicate some evidence that toxaphene may be modified based on the shape of the gas chromatographic "fingerprint."

Virtually no information seems to be available on the chemical breakdown products of toxaphene in soil and water.

Biological accumulation. Biological accumulation occurs by two processes i.e., direct absorption through body surfaces exposed to the external environment, and through the food. When natural food is involved, especially when increased concentrations of a contaminant occur through ascending trophic levels of a food chain, this accumulation is called "biological magnification."

Research shows that toxaphene is sufficiently stable to be available, in areas of regular usage, for biological accumulation if other critical requirements are satisfied; namely, that rates of uptake, metabolism and excretion are favorable for accumulation. Information is available on biological accumulation of toxaphene in warm blooded animals from feeding studies using domestic animals. Toxaphene/Strobane storage in animal fat will occur. The degree of concentration seems less than for some other chlorinated hydrocarbon insecticides and persistence of the toxaphene residues is of shorter duration (Tables 13 and 14).

Comparatively little information is available about bioaccumulation of toxaphene/Strobane in aquatic organisms. Studies indicate the presence of toxaphene residues in fish, but little information is given relating exposure rate and frequency values, and none have determined residue residence time after cessation of exposure.

Johnson and Lew (16) determined chlorinated hydrocarbon insecticide residues in fish of the Lower Colorado River system which drains on irrigated agricultural area where insecticides are often used. The following residues of DDT and its congeners, DDE and TDE, and toxaphene were reported in the fat and/or viscera of these fish.

Carp: DDT, etc., 2.0 - 185.0 mg/kg (87.0 ave.); toxaphene, 50.0 mg/kg

Channel Catfish: DDT, etc., 10.0 - 77.0 mg/kg (47.8 ave.); toxaphene, 8.2 - 11.4 mg/kg (9.8 ave.)

Sonoran Sucker: DDT, etc., 7.3 - 46.3 mg/kg (23.9 ave.); toxaphene, 2.8 - 172.9 mg/kg (32.5 ave.)

Gila Sucker: DDT, etc., 36.2 - 39.5 mg/kg (37.8 ave.); toxaphene, 25.2 - 49.9 mg/kg (34.9 ave.)

Henderson *et al*., (12, 13) reported the results of the National Monitoring Program analysis of organochlorine insecticide residues in fish collected from 50 sampling stations located in the Great Lakes and major river basins throughout the United States. Twelve toxaphene recoveries were reported from 590 composite samples taken in the fall of 1967 and spring of 1968. Concentrations ranged from 0.01 mg/kg to 1.25 mg/kg. Toxaphene was not reported in the 1969 survey. A check with the two laboratories making the analyses indicated that toxaphene was suspected in a number of samples but was not reported because of inherent analytical difficulties (11). These difficulties seem in part responsible for the relative scarcity in the technical literature of data on toxaphene in aquatic life.

Although the practice of applying toxaphene in lakes for fisheries management has been discouraged, one study of that usage revealed information on bioaccumulation in the hydrosphere under conditions of gross contamination. Terriere et al., (29) applied toxaphene in Davis Lake, Oregon at 88 μg/l in 1961 and found in 1962 and 1963 that toxaphene was present in water at average values of 2.1 μg/l and 1.2 μg/l, respectively. They reported a concentration factor of about 500 for aquatic plants, 1000 to 2000 for aquatic invertebrates, 10,000 to 20,000 for rainbow trout, 4,000 to 8,000 for Atlantic salmon and 1000 to 2000 for lake bottom mud. This lake was successfully restocked one year after treatment.

Hughes (15) has made the most complete recent study of biological accumulation in the aquatic environment in his study of toxaphene persistence in Wisconsin lakes. When applied to the lakes in fisheries management, toxaphene in the lake water declined rapidly to less than detectable amount (1 μg/liter) within 9 to 12 months. However, aquatic fauna, particularly fish stocked in the lakes following treatment, accumulated as much as 18 μg of toxaphene residues per gram of body weight. In general, prey fish accumulated higher concentrations of toxaphene than did predators. Bluegills stocked in Fox Lake about eight months following the last of 3 treatments accumulated 9.4 μg/g in 176 days and then toxaphene residues began declining until, after 787 days, 0.8 μg/g remained. Two months after fish were stocked, plankton contained 34 μg/g. Hughes believes that toxaphene was accumulated through both the food chain and directly from water.

More information is needed to evaluate the nature and significance of biological accumulation and food chain involvement, especially in aquatic life. Controlled studies will more adequately reveal the relationship of exposure to build-up in the tissues, and also indicate rates of metabolism and excretion once exposure is discontinued. Improved analytical techniques and the availability of ^{36}Cl-labeled toxaphene should make rapid acquisition of needed data possible.

SUMMARY

Toxaphene is a long-lived insecticide. Residues in soil may be detected for several months to several years, but no build-up has occurred in the soil in areas of regular usage. Microbial decomposition, photodecomposition and/or volatilization may account for major losses from the soil surface but this is not well documented. Downward migration through the soil does not normally occur to a large degree. Studies are needed to clarify the fundamental mechanisms controlling the persistence and loss of toxaphene from the soil, and to identify break-down products.

Toxaphene can be transported from the soil surface to watercourses, "dissolved" in runoff water and adsorbed on mineral and organic sediment. Concentrations reported from stream and lake water are usually less than 1 μg/l; values for bottom sediments may be several thousand times greater. There is no evidence at this time of wide-spread occurrence of toxaphene in the nation's waters comparable to the distribution of DDT and dieldrin. However, chemists are beset by analytical limitations with toxaphene not experienced with DDT and dieldrin.

Little information is available about bioaccumulation of toxaphene/Strobane in aquatic life and of food chain involvement. Toxaphene is sufficiently persistent in the physical environment to be available, in areas of regular usage, for biological accumulation if other critical requirements are satisfied; namely, that rates of uptake, metabolism and excretion are favorable for accumulation.

A study in a lake where toxaphene was applied for fisheries management suggests that biological accumulation and transfer through the food chain to higher trophic levels can occur under such conditions. However, direct application to water resulting in sustained gross exposure of aquatic organisms is not recommended. Toxaphene residues have been found in fish, but little data are available relating frequency and rate of exposure to residue concentrations, and none have determined rate of metabolism and/or excretion during exposure or after cessation of exposure These studies are needed.

TABLE 1

Toxaphene Applied to Crops vs Recovered from Soil (a)

LOCATION	LB APPLIED/ACRE			RESIDUE IN mg/kg
Lower Rio Grand Valley				
Field 1	1956-64 16.2	1965 3	1966 2	Oct 1966 2.90
Field 2	1958-64 47	1965 7	1966 1.25	Oct 1966 1.98
Field 3	1958-64 34(+1.7 Strobane)	1965 9	1966 1.	Oct 1966 1.77
Field 4	1955-64 29.25			Oct 1966 2.01
Field 5	1956-64 39.16			Oct 1966 2.43
Dade County, Fla.				
Field 1		1965 4		Mar 1968 1.21
Field 2	1958-64 8	1965 4	1966 2	Mar 1968 2.64
Field 3		1965 4		Mar 1965 0.66
Field 4	1962-64 2.2	1965 9		Mar 1968 4.14
Field 5	1962-64 31.59	1965 19.90		Mar 1968 7.00
Eastern So. Carolina				
Field 1	1952-64 3			Aug 1966 2.99
Field 2	1956-64 15		1966 3	Aug 1966 5.64
Field 3	1957-64 47	1965 9	1966 18	Aug 1966 0.99
Field 4	1956-64 38			Aug 1966 2.04

(a) Source (28)

Table 2

Toxaphene by Seasons in Flint Creek, Alabama Water (μg/l) (a)(b)

Agricultural Year	Thousand Lb. Technical Toxaphene Applied in Study Area	Summer			Fall	Winter	Spring
		Max.	Min.	Average	Average	Average	Average
1959-60	56.5	0.28	0.04	0.11	0.08	0.05	0.05
1960-61	37.9	0.41	0.01	0.21	0.06	0.02	Positive
1961-62	64.6	0.11	0.04	0.07	0.03	0.05	No Sample
1962-63	72.0	0.15	0.05	0.10	0.07	0.05	0.07
1963-64	7.5	0.08	0.04	0.16	0.03	0.01	0.01
1964-65	8.0	0.11	0.00	0.05	0.01	0.00	0.00
1965	8.5	0.08	0.00	0.01	0.00	--	--

(a) Source (25)

(b) Values are not corrected for the efficiency of the sampling and extraction methods.

Table 3

Toxaphene Concentration in California Surface Waters ($\mu g/l$) (a) (b)

Sampling Station	Max.	Min.	Average
Feather River at Nicolaus Bridge	--	--	--
American River at Sacramento	--	--	--
Sacramento River at Walnut Grove	0.40	0.03	0.10
Mokelumne River at Highway 99	--	--	0.04
Little Connection Slough at Atherton Road	--	--	0.16
Middle River at Victoria Canal	--	--	--
Delta Mendoto Canal at Head	0.12	0.03	0.08
San Joaquin River at Antioch	0.32	0.05	0.15
Suisan Bay at Martinez	0.09	0.05	0.06
Napa River at Duttons Landing	--	--	--
San Pablo Bay at Pt. San Pablo	--	--	0.08
San Francisco Bay at Berkeley Pier	0.23	0.03	0.13
San Francisco Bay at Treasure Island	--	--	--
So. San Francisco Bay at San Mateo Br.	--	--	0.26
Golden Gate Br. at Fort Point	--	--	--
San Joaquin River at Vernalis	0.93	0.02	0.26
San Joaquin River at Fremont Ford	0.46	0.04	0.13
Salton Sea near North Shore	0.40	0.05	0.14
Alamo River	0.65	0.30	0.47
All American Canal at Alamo River	0.08	0.04	0.06

(a) Source (3)

(b) Sample size 5 liters; analytical method, microcoulometric gas chromatography; sensitivity of method, 0.02 to 0.05 $\mu g/l$.

Table 4

Toxaphene in Agricultural Drains in California (µg/l) (a)

Sampling Station	Max.	Min.	Average
Reclamation District No. 108 Drain			0.23
Colusa Basin Drain			
Staten Island Drain			
Roberts Island Drain at Whiskey Slough			
Panoche Drain	5.50	0.10	1.47
Salt Slough	0.44	0.04	0.17

(a) Adapted from Baily and Hannum (3).

Table 5

Toxaphene in California San Joaquin Valley Tile Drain Effluents (ug/l) (a)

Year	Number Samples	Times Detected	Concentration Max.	Concentration Min.	Concentration Average (b)
Sept.1963-Dec.1964	16	6	0.70	0.20	0.43
1965	50	7	0.95	0.13	0.61
1966	105	17	0.88	0.21	0.37
1967	121	4	0.32	0.02	0.15
1968	79	10	0.50	0.02	0.26
1969	51	7	1.26	0.09	0.44
Totals	422	51	1.26	0.02	
		12 %			

(a) Source (1)

(b) Average of positive samples.

Table 6

Toxaphene in California Central Valley Surface Agricultural Waste Water Drains(ug/l)(a)

Year	Number Samples	Times Detected	Concentration Max.	Concentration Min.	Concentration Average (b)
Sept.1963-Dec.1964	73	40	5.50	0.04	1.02
1965	115	67	8.16	0.23	2.08
1966	89	56	7.60	0.115	1.42
1967	95	15	71.00	0.06	10.13
1968	56	27	15.00	0.11	2.47
1969	19	11	31.50	0.216	4.80
Totals	447	216	71.00	0.04	
		48 %			

(a) Source (1)

(b) Average of positive samples

Table 7

Toxaphene in California Central Valley Surface Waters (ug/l) (a)

Year	Number Samples	Times Detected	Concentration Max.	Min.	Average (b)
Sept.1963-Dec.1964	232	73	0.90	0.02	0.11
1965	158	12	0.93	0.29	0.50
1966	203	2	0.31	0.08	0.20
1967	61	0	--	--	--
1968	58	1	--	--	0.10
Totals	712	88 12 %			

(a) Source (1).

(b) Average of positive samples.

Table 8

Toxaphene in California Bay and Ocean Waters (ug/l) (a)

Year	Number Samples	Times Detected	Concentration Max.	Min.	Average (b)
Sept.1963-Dec.1964	32	7	0.26	0.03	0.12
1965	49	1	--	--	0.60
1966	51	0	--	--	--
1967	47	0	--	--	--
1968	21	0	--	--	--
Totals	200	8 4 %			

(a) Source (1).
(b) Average of positive samples.

Table 9

Toxaphene in California Sediments (ug/l) (a)(b)

Source	Max.	Min.	Average
Streams			
Feather River at Nicolaus Br.	--	--	--
Sacramento River at Walnut Grove	130	5	57
Little Connection Slough at Altherton Road	--	--	170
Middle River at Victoria Canal	--	--	--
San Joaquin River at Antioch	--	--	140
San Joaquin River at Vernalis	--	--	--
Bays			
Sunset Bay at Martinez	--	--	--
San Pablo Bay at Pt. San Pablo	--	--	110
So. San Francisco Bay at San Mateo Br.	110	88	99
Agricultural Drains			
Reclamation District #108 Drain	--	--	210
Colusa Basin Drain	--	--	--
Staten Island Drain	--	--	110
Roberts Island Drain at Whiskey Slough	--	--	380

(a) Source (3)

(b) The method of reporting concentration is unique and not relatable to ug/g of sediment in the usual manner. Concentrations are reported as parts of pesticide per parts of wet sediment. A representative location of the sample was dried and a moisture content determination was made. The pesticide concentrations were then adjusted to parts per parts of dry sediments from the relationship $Cs=\frac{100C-CwSm}{Sd}$ in which Cs=dry weight pesticide concentration in overlying water sample; Sm=percent soil moisture in sample; and Sd=percent dry material in sample.

Table 10

Insecticide Residue in Alabama Soil Samples Collected from 33 Cotton Fields (a)
(ug/kg)

Compound	Percent Positive	Mean Conc. All Samples	Mean Conc. Positive Samp.	Range
Toxaphene	58	410	710	160-1600
DDT	85	250	300	20-530
BHC	49	20	50	10-380

(a) Source (9)

Table 11

Comparison of Insecticide Recovery from Sediment and Water,
Hartselle, Alabama Water Treatment Plant (a)

Sample Source	No. Sample	Percent Positive for			
		DDT	DDE	Toxaphene	BHC
Sediment from treatment plant settling basis	45	71	64	18	22
Suspended sediment extracted from raw water by filtration prior to carbon filtration (b)	77	69	62	10	17
Carbon adsorption samples collected from water after removal by above filtration	77	13	12	31	74

(a) Source (25)

(b) A Cuno Micro-Klean filter that removed sedimentary particles larger than 25 microns was used. Smaller particles pass through to the carbon adsorption units.

Table 12

Toxaphene in Wisconsin Lakes, 1965 (a)
(parts per billion)

Lake	Year of Treatment	Treatment Rate	Water (b)	Suspended Matter	Aquatic Plants	Sediment
Little Green	1956	100	1	40	--	20
Emily	1959	100	4	20	400	200
Kusel	1960	100	3	200	70	400
Marl	1960	100	3	9	80	1000
Big Twin	1963	100 & 50	2	20	40	800
Wilson	1964 May 1965	2.5-3.5 5 Epilimnion only	4	80	50	500
Round	1964 & 1965	5 + 5	2	200	80	600
Comstock (Surface) 6.5 meters	June, 1965(c)	100	20 4	100 500	50 --	1000 --

(a) Source (17)

(b) Remaining in water after filtration through Whatman GF/A glass filters.

(c) Comstock Lake was treated 14 days prior to sampling.

Table 13

Insecticides in the Fat of Cattle after Multiple Spray Treatments (a)

Insecticide	Spray Interval (weeks)	After indicated sprayings (b) mg/kg						After last spraying mg/kg		
		1st	2nd	3rd	4th	5th	6th			
DDT, 0.5%	3	18	31		33		35	12 wks 8	24 wks 5	36 wks 2
Dieldrin, 0.5%	3	7	10	16	24			11 wks 17	28 wks 6	
Heptachler, 0.5%	2	11	14	14	20	18	19	8 wks 16	16 wks 2	
Strobane, 2%	2						29	6 wks 9	10 wks 4	14 wks 3
Toxaphene, 0.5%	2	0	0	1	7	10	6	4 wks 4	6 wks 4	

(a) Source (19).

(b) Fat samples were taken at the end of the intervals between spraying.

Table 14

Insecticide Residues Stored in the Fat of Cattle Fed Known Amounts in Their Diet (a)

Insecticide	mg/kg in Feed	mg/kg Weeks after feeding				mg/kg Weeks after feeding ceased				
		4	8	12	16	4	8	16	20	24
Aldrin	25	50	78			51	36		20	
BHC	100	159	223	230	250	84		17		
Chlordane	25	12	18			14	5		0	
DDT	25	22	34	42	40	19		11		6
Toxaphene	100	26	34	33	38	14	3			

(a) Source (19).

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TOXAPHENE RESIDUES IN ATMOSPHERIC SAMPLES

Data of toxaphene residues in atmospheric samples are very limited (1,3). Nine locations for pesticide monitoring were established at Baltimore, Md., Buffalo, N. Y., Dothan, Ala., Fresno, Calif., Iowa City, Iowa, Orlando, Fla., Riverside, Calif., Salt Lake City, Utah, and Stoneville, Miss. The identification of toxaphene was carried out by gas-liquid chromatography using two different column packings. Toxaphene identification was verified by three characteristic elution peaks on the chromatograph, one peak emerging just before p,p'-DDT and the other two after DDT (2). Further verification of the presence of toxaphene in the air samples was obtained from the person collecting the samples in Stoneville. He reported that toxaphene, DDT, and methyl parathion had been recently used at the Stoneville location (2).

Of the nine locations monitored, three showed significant toxaphene residues as follows (1):

Location	Total Number of Samples	Positive Samples	Range (ng/m^3)
Dothan	90	11	27.3-79.0
Orlando	99	9	20.0-2520.0
Stoneville	99	57	16. -1110.

Syracuse University Research Corporation under contract with NAPCA (now Research and Monitoring, EPA) has monitored pesticides in the atmosphere at six locations in New York State, one at Winter Haven, Fla., and one at Lubbock, Tex. for the past 6 mo. No toxaphene residues were present, although DDT, aldrin and endrin residues were found in some of the stations (4).

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THE EFFECT OF TOXAPHENE ON BENEFICIAL ARTHROPOD POPULATIONS

The information presented on this general subject has been separated into effects of toxaphene on pollinators and insect parasites and predators. Some excellent reviews have been published on the effect of pesticides on nontarget organisms (1, 25, 34, 39).

Effect of toxaphene on insect pollinators. The honey bee has been used in many toxicity tests because it is the most beneficial pollinating insect. The honey bee is the major agent in the pollination of most of fruit, vegetable, seed and pasture crops. The work conducted on the effect of arsenicals on honey bees, before the introduction of toxaphene, is not discussed in this review. Since the development of organochlorines, researchers have used laboratory and field observations to determine the effect of synthetic organic insecticides on pollinators. This work has been centered in Washington, California, Arizona, Texas and to some extent in other states and countries. In Texas tests were conducted to determine the effect of organochlorines on honey bees (49, 50, 51, 52, 53, 54). Toxaphene applied as a dust (20% toxaphene - 40% sulphur) was practically nontoxic producing only 5% mortality. Toxaphene sprays had little toxicity to bees when applied to cotton inside large cages while toxaphen - DDT, dusts of toxaphene, DDT, gamma BHC-DDT and chlordane killed from 8.2 to 10.4% of the bbes after eight applications.

In tests to determine the toxicity of organic insecticidal sprays to bees, the decreasing order of toxicity was gamma BHC>chlordane>DDT>toxaphene.

In another series of tests toxaphene dusts were slightly more toxic to bees than sprays. To summarize, the decreasing order of toxicity to bees of several insecticides was calcium arsenate>parathion>dieldrin>aldrin>BHC>chlordane>DDT>toxaphene. Toxaphene applied to vetch before it bloomed heavily showed promise for control of lygus bugs and pea aphids with minimum damage to pollinating insects.

Commercial applications of toxaphene to control injurious insects in alfalfa can be made without serious loss of bees (5, 7, 24, 25, 26, 27, 30). Toxaphene is low to moderate in toxicity and is not hazardous if applied when bees are not foraging. Roberts and Barnes (40) grouped pesticides according to their toxicity to bees as: (1) highly toxic, (2) moderately toxic and (3) relatively nontoxic. Toxaphene and Strobane were grouped as relatively nontoxic. In a USDA leaflet (450), toxaphene was listed as relatively nonhazardous to bees.

Todd and McGregar (43) classified the agricultural chemicals according to toxicity to bees and indicated that toxaphene was least dangerous to bees. Toxaphene, methoxychlor and sulphur were classified as materials which could be used with safety. Hocking (23) indicated that the danger of toxaphene to honey bees was very low. Todd et al (44) indicated that toxaphene was much less lethal to bees than parathion, chlordane or DDT and caused no damage to the colonies.

In laboratory tests conducted in New Zealand, the ascending order of toxicity to honey bees was toxaphene>Strobane>thiodan>diazinon (36, 37). Toxaphene and Strobane were sprayed on white clover fields early in the morning without causing bee mortality. In Canada, toxaphene was also applied to red clover without causing abnormal mortality to pollinating insects (32).

Johansen (26) indicated that toxaphene was hazardous to the alfalfa leaf cutter, but not to alkali bees. Menke (33) concluded that 15% toxaphene dust applied to blossoming alfalfa had little effect on the activity of the alkali bee.

Effect of toxaphene on insect predators and parasites. Since arthropod species tend to come to an equilibrium or "balance," removing one or more species by frequent pesticide applications may upset the balance in arthropod populations at any given time. The resurgence of pest populations after insecticide treatment is explained by (1) the reduction of natural enemies by the pesticide along with the pest, (2) favorable influences of pesticides on the phytophagus arthropods and (3) removal of competitive species (39).

The effect of toxaphene on beneficial insects has been studied by many entomologists. Soon after toxaphene became available for cotton insect control, Parcencia and Ewing (38) found that in experiments where no sulphur was added to toxaphene an increase in red spider mite populations was evident. Spider mite infestations were also present in cotton fields next to pastures where toxaphene was used to control grasshoppers (16). Where sulphur was added to the toxaphene no spider mite increases were seen. Apparently toxaphene destroyed the parasites or predators of the red spider mites, creating an environment conductive to mite increases.

an environment conducive to mite increases.

A single application of toxaphene for cotton fleahopper control reduced populations of beneficial insects; but, the populations increased in the following 3 weeks if no further applications of toxaphene were made (17, 18). After the second to fourth application of toxaphene-sulphur dust made in a regular boll weevil control program, the beneficial arthropod populations (lady beetles, flower bugs, lacewings, *Geocoris*, assassin bugs and spiders) were practically eliminated.

In laboratory tests, toxaphene at the rate of 2.5 lbs per acre killed from 84 to 85% of the spotted ladybeetle, *Ceratomegilla fuscilabris* and *Scymnus* sp., but only about 50% of the convergent ladybeetle, *Hippodamia convergens*, population (12). Results of field observations indicate that toxaphene showed a moderate to high effect on all predators in the cotton fields.

Burke (10, 11a) reported that toxaphene was less toxic to adults of *Collops balteatus*, larvae of *Hippodamis convergens* and adult *Orius insidiosus* than dieldrin or endrin. Toxaphene, dieldrin and endrin were of about the same toxicity to larvae of *Chrysopa oculata* when applied by the dipping technique. Toxaphene and dieldrin exhibited a low level of toxicity to the several insects included in these studies.

Almand (2) reported the results of observations made on three cotton fields following insecticidal treatments. Carbofuran and toxaphene treated fields contained the greatest number of predaceous insects. Attallah and Newsome (6) reported that toxaphene decreased longevity and prevented oviposition of *Coleomegilla maculata*.

Newsom and Smith (35) reported that toxaphene-sulphur (20%-40%) reduced the population of beneficial species. Severe bollworm infestations developed on a large cotton acreage which received 3 to 5 applications of either a 20% toxaphene dust or benzene hexachloride-DDT mixture for boll weevil control. Injurious bollworm infestations developed from comparatively small numbers of eggs in fields treated with chlorinated hydrocarbon insecticides.

Bartlett (8) tested 61 pesticides against 5 hymenopterous parasites and 6 coccinellids. Toxaphene was highly toxic to all species of hymenopterous parasites and showed low to medium toxicity to the coccinellids.

Van Den Bosch *et al* (46) tested the toxicity of widely used insecticides on beneficial insects in cotton and alfalfa fields of California. Insects of the following genera were included in the study: *Orius*, *Geocoris*, *Nabis*, *Chrysopa*, and *Hippodamia*. All insecticides studied were toxic to the beneficial insects to some degree but seemed to fall into three distinct groups: 1. highly toxic-parathion and toxaphene DDT combinations; 2. moderately toxic - toxaphene, endrin, and DDT; 3. slightly toxic - demeton. Considerable specificity was evident in the toxicities of the various insecticides. *Chrysopa* larvae and *Orius* sp. were relatively tolerant to the wide variety of insecticides tested.

Toxaphene - DDT spray mixtures applied at the rate of 1.3 lbs DDT and 2.6 lbs toxaphene were extremely toxic to *Hippodamia convergens*, and aphid parasite, (*Trioxys utilies*), *Geocoris* spp. *Orius* spp., *Nabis ferus* and *Sinea diadima* (42). DDT applied at the rate of 1.3 lbs per acre was nearly as toxic to the beneficial insects as the toxaphene DDT mixture.

Toxaphene applied at the rate of 2.7 lbs per acre was not as toxic as DDT and was far less toxic than the toxaphene-DDT mixture. Parathion applied at the rate of 3.6 oz per acre was comparable to toxaphene - DDT and had generally drastic effects on the beneficial species.

Harries and Valcarce (21) found that 5% toxaphene killed 32% of the Collops vittatus 12% of the Hippodamia convergens and 36% of the Colesmegilla maculata; while 5% Strobane killed 10%, 18% and 12%, respectively. These chlorinated hydrocarbons were not as toxic to these beneficial insects as the organophosphorus compounds.

Lingren et al (31) reported that toxaphene-DDT and azodrin were highly toxic to spiders. The mean numbers of all predators were significantly greater in plots treated with trichlorofon than in those treated with Azodrin®, Bidrin® and toxaphene-DDT.

Toxaphene was not as toxic to Hippodamia convergens, Orius insidiosus and Scymnus spp. as Strobane - DDT, carbaryl, trichlorfon or dicrotophos applied for cotton fleahopper control (48). Toxaphene was more toxic to spiders than Strobane-DDT mixture. About 2 weeks after the second application was made, the beneficial insect population resurged to effective predatory levels.

Wille (55) reported a large increase of Heliothis virescens occurred following treatments of DDT, BHC or toxaphene. Apparently these materials killed the beneficial insects without eliminating the pest.

Glick and Lattimore (19) found that toxaphene BHC and chlordane reduced the beneficial insects in cotton. Toxaphene was less destructive of predators than either BHC or chlordane and the addition of DDT to the chlorinated hydrocarbons increased the toxicity to predators.

Fenton (15) studied the effect of several insecticides applied to alfalfa on beneficial insect populations. Toxaphene generally reduced the beneficial insects less than parathion, endrin or demeton.

Toxaphene is only slightly toxic to bees and can be safely used in bee pastures to control injurious insects, particularly if the material is applied when the bees are not foraging.

SUMMARY

Toxaphene is highly toxic to predators and parasites of some species and low in toxicity to others. Apparently one application of toxaphene will reduce certain beneficial insects, but they usually resurge to normal levels within a few weeks. Regularly scheduled toxaphene treatments applied at intervals of 5 to 7 days generally will eliminate beneficial insect populations in crops.

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RESIDUES IN FOOD CROPS AND FOODS

Tolerances for Toxaphene Residues

The following tolerances for toxaphene residues in raw agricultural crops have been established and were in effect as of September 1971 in the United States, Canada, Germany and The Netherlands:

United States

2 ppm

Soybeans

3 ppm

Pineapples
Bananas (0.3 ppm in edible pulp)

5 ppm

Grain (Barley, oats, rice, rye, sorghum grain, wheat, cottonseed)

6 ppm

Crude Soybean Oil

7 ppm

Fruits (stone, pome, citrus, cane and strawberries)
Nuts (Hazel, hickory, pecan, walnut)
Meat Fat (Beef, sheep, goat, swine, horse)
Vegetables (Beans, black-eyed peas, broccoli, brussels sprouts, cabbage, cauliflower, carrots, celery, collards, corn, cowpeas, eggplant, green beans, horseradish, kale, kohlrabi, lettuce, lima beans, okra, onions, parsnips, peanuts, peas, peppers, radishes, rutabagas, snap beans, spinach, tomatoes)

Canada

3 ppm

Oats, rye, wheat, pineapples

5 ppm

Barley, grain sorghum, rice

7 ppm

Fruits (citrus, peas, strawberries)
Meat fat (cattle, goats, sheep, swine)
Vegetables (beans, black-eyed peas, broccoli, brussels sprouts, cabbage, cauliflower, celery, eggplant, kohlrabi, lettuce, okra, onions, peas, tomatoes)

Germany

0.4 ppm

Pears, strawberries, raspberries, cherries, plums

The Netherlands

0.4 ppm

Fruit, vegetables (except potatoes)

Residues in Food

Toxaphene is registered for a variety of uses on food crops and livestock. During 1965-1968, FDA market-basket surveys showed toxaphene to be virtually absent from these samples. The frequency of occurrence of toxaphene residues in these studies was less than that of the first 15 most commonly found pesticides. The market-basket samples represent the total diet of a 16-19 year-old male, and are obtained from retail stores in 5 regions at bi-monthly intervals. Food is prepared for consumption and analyzed for pesticide residues using gas-liquid chromatography methods.

In the later period June 1968-April 1969, toxaphene was detected in 13 of the 360 composite samples analyzed. Range of residues was 0.02 to 0.33 ppm in food categories, garden fruit, vegetables, and meat fat. DDT was the most frequently found residue being detected in 176 samples in the range of 0.003 to 0.47 ppm.

FDA surveillance studies include an annual examination of about 25,000 samples. These samples are taken objectively to characterize the pesticide residues of food shipped and consumed in the United States. They are in addition to those that are analyzed in enforcement programs designed to verify suspicions of excessive residues resulting from pesticide misuse.

A summary of the surveillance program results for toxaphene in the period 1964-67 is given below using the food categories established by FDA (see Table 1). These data reflect the widespread usage of toxaphene on vegetables and the retention of some of the residue in the processed (canned, dried, or frozen) food. Toxaphene residues were sixth most frequent in occurrence of all pesticides in processed foods, but few, if any, were in excess of the 7 ppm tolerance.

Toxaphene finds its most intensive agricultural use on cotton. It is also used to a lesser extent on other oil seed crops such as soybeans, peanuts and corn. Analysis of oil and other products derived from these crops show toxaphene is found in about 30% of the cottonseed samples, 8% of the soybean samples and 2% of the peanut samples. Above-tolerance residues have not been a problem either in the raw agricultural commodities or in the processed oils and meals. Table 2 is a summary of toxaphene residues found during 1964-1966 in oily crops.

Residues in Livestock

Toxaphene residues can be accumulated in fat of animals from ingestion and by dermal absorption. The storage level is much less than that of most other chlorinated hydrocarbon pesticides, and an equilibrium with the exposure level is rather quickly achieved. Elimination of toxaphene from the fat is quite rapid when the input is reduced. Storage-feed ratios for various animal species are summarized as follows:

	Storage-feed[(a)] ratio	Observation Period
Cattle	0.5	16 weeks
Sheep	0.3	16 weeks
Dog	0.3	2 years
Rat	0.4	2 years

(a) Storage-feed ratio $= \frac{\text{ppm in fat}}{\text{ppm in feed}}$

The rapid elimination of toxaphene residues from the fat of meat animals allows it to be used for ectoparasite control on livestock within 28 days of slaughter. Where shorter pre-slaughter intervals are required, other pesticides must be used.

USDA Meat and Poultry Inspection Programs have been established to regularly examine tissues from meat animals and poultry slaughtered in federally-inspected plants. Total number of animal tissue samples analyzed in the 27-month period from January, 1969 through March, 1971 was 7,265. Of these, only 5 contained toxaphene residues. In the same period, of 5,504 poultry samples analyzed, 2 contained toxaphene. A tabulation of these data is given in Table 3. No residue levels are given in this summary report. Only 1-2% of the samples found to contain any pesticide residues were above the tolerance limit.

Residues in Milk

Consistent with the fat-storage properties of toxaphene in livestock, transmission of toxaphene residues to milk follows the same pattern (8). Equilibrium with input is reached within about one week, and the ratio of toxaphene concentration in the feed to that in the milk is about 100:1. Excretion of toxaphene in milk declines quickly when exposure ceases.

In feeding trials, milk free of toxaphene residues was produced within 2 weeks after cessation of feeding at levels of 10 ppm. Fluid milk or dairy products do not often contain toxaphene in FDA surveillance programs. At a feeding level of 20 ppm in the daily diet for 11 weeks, toxaphene-free milk was produced 4 weeks after toxaphene-containing feed was discontinued.

Residue Decline - Controlled Studies

Table 4 is taken from the FAO-WHO monograph on toxaphene residues in food. It selectively summarizes toxaphene residue data on representative crops when normal agricultural practices are followed.

Half-lives of toxaphene residues on growing leafy crops are in the range of 5-10 days; residues from emulsifiable formulations are typically higher than those from wettable powders or dusts.

Studies of toxaphene residues on alfalfa and clover show that half-lives (corrected for crop growth dilution) are consistently in the range of 9 to 13 days under widely varying climatic conditions. Studies were conducted in Arizona, California and Delaware.

Mechanism of Residue Loss

Evaporation. Summarized in Table 5 are data comparing volatility of toxaphene with that of DDT. These measurements as well as field studies indicate that toxaphene is more volatile than DDT, and that volatility can be a significant factor in the loss of toxaphene from treated areas. Tests of toxaphene volatility from thin film on glass plates show greater loss of early-eluting GC components. Examination of field-weathered crop residues do not show evidence of such selective loss, but are similar in composition to parent toxaphene.

Toxaphene was easily washed from smooth glass surfaces by heavy rains, in contrast to deposits on crops, which are much more resistant to wash-off by rain. Sunlight had little effect on the rate of loss of thin films of toxaphene from glass plates. Half-lives of 4 days were found under conditions of indoor exposure at summer temperatures ranging to 34°C. Indoor exposure during winter months (19-24°C) revealed a half-life of 26 days. In an oven heated to 38°C, a half-life of 3 days was observed. Addition of alfalfa plant wax caused an appreciable decrease in the rate of loss at 38°C, the observed half-life being 8 days.

Attempts to detect possible toxaphene metabolites. The complex composition of toxaphene has made explicit metabolic fate studies in crops and animals impossible. Early research workers have used non-specific "total organic chloride" methods, lacking specificity, and yet accounting for all of the chlorine-containing species, whether parent compound or derived therefrom. Other methods for analyzing possible toxaphene metabolites were also unsuccessful, including paper-and thin-layer chromatography and gas-liquid chromatography.

There is no evidence for the existence of toxaphene conversion products in weathered crop residues or in fat deposits from animals exposed or fed with toxaphene. Carter *et al*. (3) examined weathered toxaphene residues on alfalfa and found insecticidal activity was the same as that of toxaphene. Residues in fat of steers wintered on toxaphene-treated alfalfa hay were similar in infrared absorption and insecticidal activity as authentic toxaphene.

Klein and Link (6) examined residues on toxaphene-sprayed kale and found that over 99% of the original residue was lost during the first two weeks. Gas chromatographic analysis of the residues indicated a modest loss of early-eluting GLC components. However, the composition of the residue even after 4 weeks was readily recognizable as toxaphene from the GLC elution pattern.

Carlin (2) concluded that no toxaphene conversion products were formed in alfalfa treated with toxaphene and allowed to weather. These conclusions were based on "total chloride" methods, electron capture GLC, and bioassays, the last showing no greater toxicity than authentic toxaphene.

Possible metabolites of toxaphene. Attempts to introduce functional groups into toxaphene by *in vitro* chemical reaction have been unsuccessful, and the availability of model compounds as authentic reference standards for various separation and detection systems has been limited.

Recently, samples of "keto-toxaphene" and "hydroxy-toxaphene" were prepared by Buntin. (1) Camphor was chlorinated to a value corresponding to the addition of 7 atoms of chlorine. The resulting "keto-toxaphene," a viscous pale yellow liquid, was reduced with lithium aluminum hydride to form "hydroxy-toxaphene." These compounds are less toxic to flies and rats than toxaphene; gas chromatography shows they elute with the early peaks of toxaphene.

Cleanup techniques applied to keto-toxaphene and hydroxy-toxaphene show that the former survives fuming sulfuric acid, but that hydroxy-toxaphene does not. Dehydrohalogenation (as applied to toxaphene prior to gas chromatography) showed that these compounds are retained in the alkaline aqueous phase when it is extracted with hexane. Both compounds are extracted by hexane from distilled water.

Weathered toxaphene residues from alfalfa were examined for the possible presence of keto-toxaphene or hydroxy-toxaphene. No evidence for their presence was found (2).

Metabolism in the Honey Bee

A study of toxaphene residues in rape oil, honey, and bees was conducted by Jumar and Sieber (5). They prepared a ^{36}Cl-tagged toxaphene and determined that residues were transmitted to rape oil in the range of 0.3 to 1.5 ppm, depending on the method of application to the rape plant. Honey made by bees exposed to the toxaphene-treated rape plants contained less than 0.01 ppm toxaphene. The study on toxaphene in the bee employed ^{82}Br-toxaphene (one Cl atom replaced by ^{82}Br). More than 95% of toxaphene absorbed by bees from feeding was stored briefly in the body before release as a chlorine-containing water-soluble compound which was not identified.

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TABLE 1

DOMESTIC FOODS SURVEILLANCE BY FDA -- 1964 to 1967

	Toxaphene Residues	
Food Category	Incidence Percent	Average ppm
Large Fruits	0.3	T*
Small Fruits	1.3	0.01
Grains and Cereals for human use	0.3	T
Leaf and Stem Vegetables	6.4	0.18
Vine and Ear Vegetables	1.4	0.01
Root Vegetables	1.1	T
Beans	0.9	T
Eggs	0.2	T
Nuts	0.3	T
Processed Foods	5.0	0.45
Grains (animal)	0.1	T
Fluid Milk (fat basis)	-	-
Dairy Products (fat basis)	-	-

T* = < 0.005 ppm (trace)

TABLE 2

Summary of Toxaphene Residues
In Oil Seeds, Oils, and By-Products (1964-66)

	Toxaphene	
	Incidence	Average
	Percent	ppm
SOYBEANS	8.0	0.004
Crude Oil	4.1	0.024
Meal (cake)	-*	
Refined Oil	4.3	**T
PEANUTS	1.7	0.006
Crude Oil	2.8	0.008
Meal (cake)	-*	
Refined Oil	-*	
COTTONSEED	30.4	0.023
Crude Oil	1.3	0.010
Meal (cale)	1.1	0.003
Refined Oil	12.2	0.140
CORN GRAIN	-*	
Crude Oil	-*	
Refined Oil	-*	

* Signifies not detected
**T Signifies less than 0.001 ppm

TABLE 3

Chlorinated Pesticide Residues

In Meat and Poultry 1969-1971

(Frequency of a specific residue in animal and poultry tissue)

	Animal			Poultry		
Pesticide	1969	1970	3 mo. 1971	1969	1970	3 mo. 1971
Aldrin	14	66	2	6	51	0
BHC	523	610	59	294	517	14
Chlordane	2	2	1	0	0	0
Dieldrin	1,336	1,549	219	1,639	2,270	138
DDT + metab.	2,671	2,835	402	2,187	2,850	299
Endrin	27	104	18	87	111	29
Heptachlor	752	1,006	61	313	877	54
Lindane	505	425	34	197	242	13
Methoxychlor	74	39	13	28	27	0
Toxaphene	2	3	0	2	0	0
Total with residue	2,907	3,238	473	2,181	2,951	303
Total Samples	3,169	3,528	568	2,199	2,999	306
Total over limits	35	55	4	10	33	2

TABLE 4

Toxaphene Residues Resulting From Supervised Trials

	Rate of Application (kg/ha)	No. of Treat- ments	Pre-harvest* Interval (days)	Residue at Harvest (ppm)	Comments
Vegetables					
Lettuce	5.5	1-1	10	5.8-7.9	whole head
Kale	5.0	4	36	3.3-7.2	
Cabbage	1.9-12	2-6	9-38	0.8-6.6	on outer leaves
Spinach	5.0	4	30	16.7-18.8	
Celery	1.1-1.6	9	13	1.8 stalks 6.5 leaves	washed
Cauliflower	3.8	1	8	1.1	(processed commercially & frozen before analysis)
Broccoli	10	1	8	3.4	
Tomatoes	1.3-2.5	8-9	5-7	2.0-4.3	
Greenbeans	7.5	1	7	1.3	unwashed
Lima Beans	3.9	1	14	0.3	shelled beans
Carrots	25	2-4		0.9-3.3	soil applic. 1 yr.
Potatoes	0.95-2.5	6	21	0 detected	
Field Peas	2.5	3	4	1.8	
Oil Seeds					
Cotton (seed)	3.9-5.0	15	6	3.6-5.2	lint bearing seed
Soybeans	3.8	3	60	0.5	
Peanuts (shelled)	25-50	1		0 detected	soil treat.
Fruit					
Oranges	5.7	2	7-70	0-10.9 skins 0-0.3 pulp	
Bananas	3.8	1	1	0.3-1.3	whole fruit
Pineapple	2.8	2	81-96	1.3-2.7	whole fruit
Cereal Grains					
Wheat	1.9-3.8	1	14-21	0.5-1.8	
Barley	1.9-3.8	1	7-28	0.7-14.2	
Oats	1.9-3.8	1	7	1.0-2.6	
Rice	1.9-3.8	1	7-28	1.5-5.6	unfinished grain
Sorghum	2.5	1	28	2.5-3.1	
Corn (maize)	2.5	1	12	0.08 kernals	
Fat of Meat Animals					
Beef	0.5%	12	28	5.0	12 weekly sprays
Swine	0.5%	2	28	0-0.6	2 sprays
Shelled Nuts					
Almonds	4.0	3	136	1.5	

*Interval from last application if multiple applications were made.

Table 5

Evaporation Rates -- DDT(7) vs. Toxaphene(4)

Conditions	$\mu g/cm^2/hr$		lbs/acre/year	
	DDT	Toxaphene	DDT	Toxaphene
Room Temperature - no sunlight	2×10^{-3}	5×10^{-3}	1.6	3.9
Outdoors	3×10^{-4}	6.4×10^{-3}	0.2	5.0
Outdoors - 20°C, 10 mph wind	1×10^{-3}		0.8	
Framglass, summer			2.0	
Framglass, winter			0.3	
32-38°C, (oven) - toxaphene alone		11.9×10^{-3}		9.3
32-38°C, (oven) - toxaphene + alfalfa wax		1.7×10^{-3}		1.3

Table 6

Comparison of Toxicity of Toxaphene with Hypothetical Metabolites

Housefly Bioassay

Compound	LC_{50}, %	Toxicity Ratio
Toxaphene standard	0.052	1
Keto-toxaphene	0.17	1/3
Hydroxy-toxaphene	0.32	1/6

Rat Toxicity

Compound	LD_{50}, mg/kg	Toxicity Ratio
Toxaphene standard (a)	120	1
Keto-toxaphene	425	1/4
Hydroxy-toxaphene	>1,080	>1/9

(a) From earlier test

TOXICOLOGY IN MAN AND ANIMALS

Acute toxicity and pharmacological actions. Acutely toxic doses of toxaphene produce effects that are typical of the chlorinated hydrocarbon insecticides (1, 5, 12). Symptoms include salivation, spasms of the leg and back muscles, nausea, vomiting, hyperexcitability, tremors, chronic convulsions and tetanic contractions of all skeletal muscles.

Most of these effects are the results of diffuse stimulation of the cerebrospinal axis. After lethal doses the convulsions continue until death occurs. Respiration is arrested due to tetanic muscular contractions and then increases in amplitude and rate as the muscles relax (17, 18).

Toxic symptoms begin within an hour and death occurs in 4 to 8 hours, but may be delayed as long as 24 hours after lethal doses. The pathological changes in acute toxaphene poisoning consist of petechial hemorrhages and congestion in the brain, lungs, spinal cord, heart and intestine. Pulmonary edema and focal areas of degeneration in the brain and spinal cord are also present.

The basic mechanism responsible for the toxicity of toxaphene is unknown since no studies on this aspect of the toxicology of toxaphene have been reported. However, due to the close similarity between the pharmacological actions of toxaphene and DDT, it seems likely that findings made on the action of DDT will be applicable to toxaphene. The similarity between the pharmacological actions of toxaphene and DDT is substantiated by the fact that phenobarbital and other barbiturates effectively treat acute poisoning by both compounds.

The pharmacological actions of toxaphene and its mammalian toxicity have been known for almost 20 years. As a result, the references commonly used as sources of information for diagnosis and emergency treatment of pesticide poisoning (4, 10, 11, 20) contain essential information needed to prevent and treat toxaphene poisoning.

The acute toxicity of toxaphene was measured in a number of species. A comparison of the oral and dermal toxicity of several chlorinated hydrocarbon insecticides in rats under standardized conditions was published by Gaines (8). Table I contains data from that report. For oral administration, the compounds were dissolved or suspended in peanut oil and for dermal application xylene solutions were used.

TABLE 1

Acute Oral and Dermal LD_{50} Values for Toxaphene and Other Chlorinated Hydrocarbons to Rats (a)

Compound	Oral LD_{50} (mg/kg) Males	Oral LD_{50} (mg/kg) Females	Dermal LD_{50} (mg/kg) Males	Dermal LD_{50} (mg/kg) Females
Toxaphene	90	80	1075	780
DDT	113	118	-	2510
Chlordane	335	430	840	690
Aldrin	39	60	98	98
Dieldrin	46	46	90	60
Endrin	18	8	-	15

(a) Data from Gaines (8)

The data in Table 1 show that toxaphene resembles DDT in acute oral toxicity to rats but is more toxic by single dose dermal application than DDT. A number of factors influence the toxicity of toxaphene. The route of administration, the solvent used for the tests, and the species must be considered in evaluating the potential hazard. Information on the influence of these factors on the toxicity of toxaphene was obtained by compiling the acute toxicity data in the literature.

The data in Table 2 show the range of variation in toxicity of toxaphene given orally to several common species.

Table 2

Acute Toxicity of Toxaphene (a)

Species	Route	LD_{50} (mg/kg)	Vehicle
Rat	oral	90	peanut oil
Rat	oral	60	corn oil
Rat	oral	120	kerosene
Mouse	oral	112	corn oil
Dog	oral	49	corn oil
Dog	oral	>250	kerosene
Guinea pig	oral	270	corn oil
Guinea pig	oral	365	kerosene
Cat	oral	25-40	peanut oil
Rabbit	oral	75-100	peanut oil
Rabbit	oral	250-500	kerosene
Cattle	oral	144	grain
Goat	oral	200	xylene
Sheep	oral	200	xylene
Rat	dermal	930	xylene
Rabbit	dermal	>4000	dust
Rabbit	dermal	< 250	peanut oil

(a) Bulletin by Hercules Incorporated (12).

Assuming man resembles the most sensitive experimental species, the lethal dose for a 70 kg adult would be around 2 to 3.5g. The fatal dose for man was estimated to be from 2 to 7g (1, 4, 10).

Acute toxaphene poisoning in humans is rare. When this material was first used (17), four cases of poisoning by ingestion in children under 4 years of age were reported occurred. The same report contained a description of severe toxaphene poisoning in adults following misuse of the pesticide in agriculture. The quantity of toxaphene estimated to have been ingested by three of the people ranged from 9.5 to 47 mg/kg.

Due to the long period of use and experience with toxaphene and its moderate toxicity, accidental poisoning by this insecticide is now extremely uncommon. In contrast, accidental poisoning by possible substitutes such as the organophosphorus insecticides are expected to exceed those that resulted from toxaphene because of the higher toxicity of the organophosphates.

Inhalation of toxaphene can cause irritation of the respiratory tract. Warraki (22) has described acute bronchopneumonia with miliary shadows in two men with an occupational history of heavy and prolonged exposure to toxaphene sprays. The threshold limit value for atmospheric levels of toxaphene has been established at 0.5 mg per cubic meter of air (7).

Subacute toxicity. The subacute toxicity of toxaphene was studied by Ortega *et al.*, (19) in small groups of rats fed 50 and 200 ppm in the diet. These dietary levels produced no clinical signs of toxicity or inhibition of food consumption or growth rate. Only the livers, spleens and kidneys were examined histologically. There was no damage to the kidney or spleen but the livers of 3 of 12 rats that received 50 ppm showed slight liver changes. Six of 12 rats fed 200 ppm showed distinct liver changes.

A subacute toxicity study on dogs was done (16) in which two dogs received 4 mg/kg (about 160 ppm) for 44 days and two other dogs received the same dose for 106 days. There was occasional central nervous system stimulation for a short time after administration. Degenerative changes in the kidney tubules and liver parenchyma were seen.

Cattle and sheep were fed toxaphene at concentrations as high as 320 ppm in the diet for 134 and 151 days. At the highest level (320 ppm) two steers showed central nervous system stimulation with tremors. There was no hematological or pathological changes in the tissues.

Chronic toxicity. The chronic toxicity of toxaphene has been studied in rats using the conventional 2-year feeding period at levels of 25, 100 and 400 ppm in the diet (6). Only the liver showed significant changes at the 100 and 400 ppm levels.

In dogs fed 40 ppm of toxaphene in the diet for 2 years there was slight degeneration of the liver, and at 200 ppm moderate degeneration of the liver occurred (21). There were no liver changes in groups of 2 dogs fed 5, 10 or 20 ppm of toxaphene for 2 years (2).

Reproduction, teratology and mutagenesis. A three-generation reproduction study was conducted on rats fed 25 and 100 ppm toxaphene (14). This study was carried out using the currently accepted protocol with respect to numbers of animals and the types of measurements that were made.

There were no differences between control and toxaphene-treated rats in reproductive performance, fertility, lactation, or the viability, size and anatomical structure of progeny.

An earlier study was done on pheasants fed 100 and 300 ppm of toxaphene (9). The 300 ppm level caused a decrease in egg laying and hatchability and in the food intake and weight gain. Both dose levels caused greater mortality in young pheasants during the first 2 weeks after hatching than was observed in the controls.

No evidence of a carcinogenic action by toxaphene was obtained in the chronic toxicity studies described above. A recent experiment (13) was conducted to detect turmorigenicity of pesticides by oral administration of maximum tolerated doses to mice starting at 7 days of age and continuing to 4 weeks of age. From 4 weeks of age until 18 months of age, the chemicals were fed in the diet at levels near the maximum tolerated dose. Toxaphene was not included in that study but the closely related material, strobane, given at a daily dose of 4.64 mg/kg caused a higher incidence of lymphomas than was seen in controls. No studies on the possible mutagenic effects of toxaphene have been reported.

Interactions. Toxaphene can change the toxicity of drugs and other chemicals detoxified by hepatic microsomal enzymes and alter steroid metabolism because it induces synthesis of hepatic microsomal enzymes (15). Dose-response relationships for enzyme induction by toxaphene were measured by feeding various dietary levels to rats for 13 weeks. The lowest dietary level of toxaphene that cause induction of one or more of the three microsomal enzyme systems studied was 5 ppm.

Maximum induction occurred within the first 3 weeks of the feeding period at all levels of toxaphene that cause enzyme induction. After this time the activity was maintained at a constant elevated level until feeding of the pesticide was discontinued.

These results show that levels of 5 ppm and higher could alter the metabolism rate of other chemicals. Similar enzyme induction was obtained with DDT at a dietary level of 1 ppm. No similar quantitative measurements of dose-response relationships for enzyme induction with toxaphene were conducted on other species.

Except for interactions caused by enzyme induction, there have been no studies showing any other type of interactions that could be caused by toxaphene.

Tissue residues. Toxaphene accumulates in the fat of man and animals. With any given rate of subacute intake, a certain storage level is attained with no build up above this level, and when the intake of toxaphene is stopped the residue rapidly decreased (3).

The storage level of toxaphene is lower and elimination is more rapid than with most other chlorinated hydrocarbons. In cattle and sheep the storage level in fat is one-fourth to one-half of the level in the feed. The storage level in fat of hogs is somewhat less than in other livestock probably because of the greater total fat content. No residue studies were reported on human tissues. Future analysis of autopsy material for pesticide levels should include toxaphene.

Summary

The mammalian toxicity of toxaphene was measured in various experimental animals. Since toxaphene was one of the earliest chlorinated hydrocarbons intoduced into widespread use, the toxicity studies conducted over 20 years ago are summarized in most of the common references on the toxicity, diagnosis and treatment of poisoning by pesticides.

A few cases of fatal accidental poisoning from ingestion of toxaphene occurred during the early period of the practical use of toxaphene. Evaluation of the acute toxicity of toxaphene and its pharmacological actions is adequate as it resembles other chlorinated hydrocarbon insecticides in many respects.

Measurements of the subacute and chronic toxicity of toxaphene in experimental animals revealed that repeated high doses cause central nervous system excitation and liver injury. The latter effect occurs at lower doses fed to animals over a prolonged period. However, no liver injury occurred when rats were fed 25 ppm of toxaphene or when dogs were fed 20 ppm of toxaphene for two years.

The conventional three generation rat reproduction study showed no adverse reproductive or congenital effects by toxaphene in this species at dietary levels of 25 and 100 ppm. Egg production and hatchability decreased in pheasants fed 300 ppm and at this level, as well as 100 ppm, there was greater mortality of young pheasants during the first 2 weeks after hatching.

The conventional 2-year feeding studies in rats and dogs showed no evidence that toxaphene is carcinogenic. However, a different type of exposure in which young mice were treated from 7 days to 18 mo of age with a maximum tolerated dose indicated that Strobane caused a higher incidence of lymphomas than was seen in control mice. Toxaphene has not yet been tested for mutagenicity.

The pattern of uptake and storage of toxaphene in animal tissues is biochemically similar but quantitatively different from most other chlorinated hydrocarbons. The level of uptake is lower and the rate of elimination more rapid than with most other chlorinated hydrocarbons.

The metabolism of toxaphene, including the use of isotope-labeled material, has received very little attention. Most investigators are reluctant to study a substance that is a mixture of related compounds rather than a single chemical agent.

Toxaphene causes induction of hepatic microsomal enzymes when dietary levels of at least 5 ppm are fed to rats. There is no evidence that toxaphene could change the toxicity of other chemicals through any mechanism other than enzyme induction.

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TOXAPHENE RESISTANCE
IN ARTHROPODS

Some insects have always been able to survive the most effective insecticidal treatments that man has been able to devise. Insect resistance to insecticides was first realized in 1908, when the San Jose scale developed resistance to lime-sulphur in the State of Washington. The term resistance is used here to describe an insect population which consistently exhibits greater survival from repeated exposures to a chemical insecticide than was noticed when the chemical was first used (16). The World Health Organization Expert Committee on Insecticides (7), proposed the following definition:

> "Resistance to insecticides is the development of an ability in a strain of insects to tolerate doses of toxicants which would prove lethal to the majority of individuals in a normal population of the same species. The term "behavioristic" resistance describes the development of the ability to avoid a dose which would prove lethal."

The "behavioristic resistance" concept is associated with the feeding preferences or the avoidance of a chemical deposit. For example, certain mosquitoes may move away from an insecticidal before absoring a lethal dose or they may not remain on an insecticidally treated surface long enough to be poisoned before being stimulated to fly away. Thus, they have developed behavioral traits which prevent them from being poisoned by certain chemicals.

There are two types of resistance to chlorinated hydrocarbon insecticides: (1) to DDT and its analogues, and (2) to the cyclodiene derivatives such as dieldrin, chlordane, toxaphene and gamma-BHC. Insects, made DDT-resistant with DDT selection pressure are cross-resistant to DDD, and resistant to methoxychlor and perthane, but not the cyclodiene derivatives, toxaphene or BHC. Insects made dieldrin-resistant by dieldrin selection pressure are cross-resistant to the other cyclodiene derivatives and to BHC, but not to DDT and its relatives. Cyclodiene-resistant strains of insects are cross-resistant to gamma BHC, and gamma BHC-resistant insects are cross-resistant to cyclodienes. Cyclodiene-resistant strains are apparently always resistant to toxaphene (7, 8).

According to Brown (8), all cases of resistance to dieldrin and other cyclodiene derivatives, and to BHC involves resistance to toxaphene also. Where the word toxaphene or dieldrin is listed under insecticide (Table 1), it is because this type of cyclodiene-BHC-resistance was induced under toxaphene or dieldrin pressure.

Several authors (5, 6, 7, 8, 10, 19, 20, 23) have reviewed the literature on insect resistance. The mechanism of resistance to cyclodiene derivatives such as dieldrin, endrin and heptachlor still is unknown. The well-known cyclodiene-resistant strains of the boll weevil do not absorb less dieldrin than nonresistant strains and apparently do not detoxify this compound.

The nerves of cyclodiene-resistant flies refract high levels of dieldrin, which means the composition of the ganglia may be crucial for developing this kind of resistance. The excretory activity of the Malpighian tubules is inhibited by dieldrin. Cyclodiene-resistant flies continue to excrete dieldrin long after susceptible flies have ceased activity, but this may be a consequence, not a cause.

Some BHC-resistant flies absorb gamma BHC at a lower rate than normal resistant flies. However, the lower rate of absorption and higher detoxification does not fully explain BHC-resistance. As with cyclodienes, resistance apparently resides in the ganglia themselves.

Insecticide-resistant pests are a problem in both agricultural and medical entomology. Melander (21) is generally credited with publishing the first report of an insect developing resistance to an insecticide. In field experiments conducted at several locations in the State of Washington, Melander found that San Jose scale was resistant to lime-sulphur at Clarkston. Flint also (12) reported the same findings in Illinois. The examples of resistance to lime-sulphur, lead arsenate, hydrogen cyanide, phenothiazine and tarter emetic are discussed in the reviews.

In 1946, 2 years after the introduction of DDT, housefly resistance to this compound was demonstrated in Italy and Sweden. In 1947, DDT resistance in the housefly appeared in Egypt and New York and in 1948 was reported in many state in the United States. Resistance of the housefly to DDT led many scientists to study the physiology, mechanism and genetics of resistance. Many of these studies included the other organochlorines. Most scientists agree that cyclodiene-resistance is completely separate from DDT resistance. By 1962, the housefly was resistant to the BHC-dieldrin group in the United States, Scandinavia, South America, Africa, USSR, Japan, India, Caribbean and Romania.

It is estimated that DDT-resistance developed in the housefly two years after the introduction of DDT, and cyclodiene-resistance usually develops within one year after the substitution of BHC or dieldrin.

Thirty-six species of Anopheles are resistant to dieldrin in various countries of the world. Twelve species of Culicine mosquitoes are resistant to the BHC-dieldrin group of insecticides (13).

DDT-resistance first appeared in the body louse during 1951 in Korea (11). At this time, toxaphene gave complete kill of the DDT-resistant lice, indicating they were not resistant to toxaphene. Later, BHC-resistant strains appeared in Japan that were also resistant to toxaphene. Malathion is now used to control the BHC-resistant lice.

Seven species of ticks are resistant to the BHC-dieldrin group of insecticides; (8, 34, 36) two of these species are found in the United States.

The boll weevil developed resistance to the chlorinated hydrocarbon insecticides, i.e., endrin, heptachlor, dieldrin and toxaphene (26, 27) in 1955. Soon after the first report of chlorinated hydrocarbon-resistance in the boll weevil in Louisiana, resistant weevils were reported in Texas (31), in most of the other cotton growing states where weevils occur (4, 17, 24), and in Mexico and Venezuela (8).

Cyclodiene resistance now has been reported in at least 18 species of insects that attack cotton. This list includes: boll weevil (17, 27, 31); bollworm (2); tobacco budworm (1, 2); cabbage looper (4); cotton leafworm (4); cotton fleahopper (24); Lygus sp. (3, 22); thrips spp. (25, 30); and salt-marsh caterpillar (29, 32). According to Brown (8) there also has been a marked increase in the past five years in the number of cyclodiene-resistant species of tobacco, rice and stored products insects.

Toxaphene-resistance in the Egyptian cotton leafworm had almost as drastic an effect on cotton production in Egypt as the development of insecticide resistance in the boll weevil in the U. S. BHC-resistance of the sugar-cane borer in Trinidad and the rice stem borer in Japan has also caused similar crises in agricultural production.

The resistance to aldrin, dieldrin and heptachlor in soil insects has become widespread. Three species of wireworms are resistant to cyclodiene insecticides. Dieldrin resistance in the onion maggot, cabbage maggot and carrot rust fly is now widespread and has increased in the seed corn maggot and turnip maggot. Insecticide resistance also has been developed by four species of Diabrotica root worms, the alfalfa weevil and white fringed beetle.(7)

Summary

Cyclodiene-resistant strains of insects are cross-resistant to gamma BHC and gamma BHC-resistant insects are cross-resistant to the cyclodienes. Cyclodiene-resistant strains are apparently always resistant to toxaphene.

Among the 149 insect species that have developed resistance to toxaphene, BHC, organochlorine insecticides and cyclodiene derivatives, 65 are of agricultural importance and 84 of public health or veterinary importance. The list of resistant agricultural pests include such important crop pests as the boll weevil, bollworm, tobacco budworm, cabbage looper, cotton fleahopper, rice steam borer and others; while the list of resistant public health pests include 26 species of Anopheles and 12 species of culicine mosquitoes as well as many ticks, flies, lice, roaches, etc.

Table 1.

Tabulation of Pests Reported to be Resistant to Toxaphene, BHC, Organochlorine Insecticides and Cyclodiene Derivatives (a).

Pest	Insecticides	Location
	Cotton	
Beet armyworm Spodoptera exigua	Organochlorine compounds	Ariz., Ark., Calif., Miss.
Boll weevil Anthonomus grandis	Organochlorine compounds	Ala., Ark., Geo., La., Miss., N.C., Oklah., S.C., Tenn. Tex., Mex., Venezuela
Bollworm Heliothis zea	Toxaphene - DDT	Texas
Cabbage looper Trichoplusia ni	Organochlorine compounds	Ala., Ark., Calif. La., Miss., Okla
	Endrin and Toxaphene	Ariz.
Cotton leafworm Alabama argillacea	Organochlorine compounds	Ark., La., Tex., Venezuela, Colombia
Spodoptera littoralis	Toxaphene	Egypt, India
Lygus hesperus	Toxaphene	Calif.
Salt marsh caterpillar Estigmene acraea	Toxaphene, DDT Endrin	Ariz., Calif.
Stink bug Euschistus conspersus	Organochlorine compounds	Calif.
Frankliniella occidentalis	Organochlorine compounds	Texas
	Toxaphene	New Mexico

Pest	Insecticides	Location
Thrips tabaci	Organochlorine compounds	Texas
Anomis texana	Toxaphene	Peru
Tobacco budworm Heliothis virescens	Strobane plus DDT	Texas
	Toxaphene plus DDT	Texas
	Endrin	La., Miss., Tex.,
Cotton fleahopper Pseudatomoscelis seriatus	Chlorinated hydrocarbons	S.E. USA
Cotton leaf perforator Bucculatrix thurberiella	Chlorinated hydrocarbons	Calif.
Cotton aphid Aphis gossypii	BHC	S.E. USA
Spiny bollworm Earias insulana	Endrin	Israel, Spain
Cotton stainer Dysdercus peruvianus	BHC	Peru
	Sugarcane	
Sugarcane Froghopper Aeneolamia varia	BHC	Trinidad
Sugarcane borer Diatraea saccharalis	Endrin	La.
	Tobacco	
Tomato hornworm Protoparce sexta	Endrin	S.C., N.C.
Dark sided cutworm Euxoa messoria	Dieldrin	Ont.
Sandhill cutworm Euxoa detersa	Aldrin	Ont.

Pest	Insecticides	Location
Potatoe tuber moth *Phthorimaea operculella*	Endrin	Queensland
	Rice	
Rice leaf beetle *Lema oryzae*	BHC	Japan
Rice stem borer *Chilo suppressalis*	BHC	Japan, Taiwan
Rice water weevil *Lissorhoptrus oryzophilus*	Aldrin	Ark., La., Miss., Tex.
Smaller brown plant hopper *Delphacodes striatella*	BHC	Japan
Rice paddy bug *Leptocoris varicornis*	BHC, Endrin	Ceylon, Thailand
Black rice bug *Scotinophora lurdia*	BHC	Taiwan
	Stored Products	
Red flour beetle *Tribolium castaneum*	BHC	Kenya
Rice weevil *Sitophilus oryzal*	BHC	England, Queensland
Granary weevil *Sitophilus granarius*	BHC	S. Africa
Maize weevil *Sitophilus zeamais*	BHC	Kenya
	Miscellaneous	
Black cutworm *Agrotis ypsilon*	Aldrin	Brazil, Taiwan
Singhara beetle *Galerucella birmancia*	BHC	N. India
Chinch bug *Blissus pulchellus*	BHC	Panama

Pest	Insecticides	Location
Coca capsid *Distantiella theobroma*	BHC	Ghana, Nigeria
Wooly apple aphid *Eriosoma lanigerum*	BHC	Queensland
Pear psylla *Psylla pyricola*	Dieldrin	Washington
Brown coca capsid *Sahlbergiella singularis*	BHC	Nigeria
Citrus thrips *Scirtothrips citri*	Dieldrin	Calif.
Banana tree weevil *Cosmopolites sordidus*	Dieldrin	Guinea, Ivory Coast, Cameroun
Tuber flea beetle *Epitrix tuberis*	Dieldrin	B.C.
Potato beetle *Leptinotarsa decomlineata*	BHC	Europe
Strawberry aphid *Chaetosiphon fragaefolii*	Endosulfan	Wash.
Serpentine leaf miner *Liriomyza archboldi*	Aldrin	Florida
	Soil Insects	
Southern potato wireworm *Conoderus fallii*	Chlordane	S.C.
Tobacco wireworm *Conoderus vespertinus*	Dieldrin	N.C.
Sugarbeet wireworm *Limonius californicus*	Aldrin	Washington
Western corn rootworm *Diabrotica virgifera*	Aldrin	Nebr., Kan., S.D. Iowa, Mo., Minn.
Northern corn rootworm *Diabrotica longicornis*	Aldrin	S.D., Ohio, Ill.,

Pest	Insecticides	Location
Southern corn rootworm *Diabrotica undecimpunctata*	Aldrin	N.C., Va.
Banded cucumber beetle *Diabrotica balteata*	Aldrin	La., S.C.
Alfalfa weevil *Hypera postica*	Heptachlor	Utah, Mont., Wyo., Nev., Calif., Va., Md., N.Y., Pa., Del., N.C.,
White fringed beetle *Graphognathus leucoloma*	Dieldrin	Ala.
Onion maggot *Hylemya antiqua*	Dieldrin	Wis., Mich., Ont., Wash., Ore., B.C., Ill., N.Y., Man. Que., Minn., Me., Ohio, France, Holland, Japan
Bean seed maggot *Hylemya liturata*	Dieldrin	Ont., Conn., Que., Nfld.
Cabbage maggot *Hylemya brassicae*	Dieldrin	Ill, Wis., Wash., B.C., Que., Nfld. Ont., Eng., N.Y., N.S., Maine, Pa., Ohio, Belgium, Germany, Sweden
Seed corn maggot *Hylemya platura*	Dieldrin	B.C., Ont., Japan, England
Turnip maggot *Hylemya floralis*	Heptachlor	Saskatchewan, Germany, Norway
Barley fly *Hylemya arambougi*	Dieldrin	Kenya
Spotted root maggot *Euxesta notata*	Dieldrin	Ont.
Large blub fly *Merodon equestris*	Aldrin	England
Carrot rust fly *Psila rosae*	Dieldrin	Ore, B.C., Ont., Wash., France, Holland

Pest	Insecticides	Location
Public Health and Veterinary Importance		
Body louse *Pediculus corporis*	BHC, dieldrin	France, Japan, West Africa, South Africa, Iran, India, Korea, Tanganyika, Sudan
Lingonathus africanus and *Lingonathus stenopsis*	BHC, dieldrin	South Africa
Cattle sucking louse *Haematopinus eurysternos*	BHC, dieldrin	Alberta
Goat biting louse *Boophilus limbata* and *Boophilus caprae*	BHC, dieldrin	Texas
Oriental cockroach *Blatta orientalis*	BHC, dieldrin	Germany, Czechoslovakia
German cockroach *Blattella germanica*	BHC, dieldrin	Texas, S.E. U.S.A. N.E. USA, Calif., Panama, Cuba, Puerto Rico, Canada, Trinidad, Japan, Poland, England, Germany Denmark, Hawaii, Australia, New Guinea

Pest	Insecticides	Location
Periplanta brunnea	BHC, dieldrin	Florida
Bed bug *Cimex lectularius*	BHC, dieldrin	Italy, Israel, Indonesia, Zambia, Rhodesia, Borneo, S. India, S. Africa, N. India, Egypt
Tropical bed bug *Cimex hemipterus*	BHC, dieldrin	West India, Tanganyika, Kenya, Haute, Volta, Dahomey, Zanzibar, Malaya, Gambia, Malagasy, S. India
Human flea *Pulex irritans*	BHC, dieldrin	Tanganyika, Turkey, Egypt
Dog and cat flea *Ctenocephalides canis* and/or *Ctenocephalides felis*	BHC, dieldrin	USA, Hong Kong, Hawaii, Japan
Oriental rat flea *Xenopsylla cheopis*	BHC, dieldrin	W. India, S.E. India Thailand
Xenopsylla astiu	BHC, dieldrin	S. India
Blue tick *Boophilus decoloratus*	BHC, dieldrin	Cape Province, Transvaal, Northern Rhodesia
Cattle tick *Boophilus microplus*	BHC, dieldrin	Queensland, Brazil, N. India, Guadeloupe Madagascar
Lone star tick *Ambloyomma americanum*	BHC, dieldrin	Okla., Madagascar
Brown dog tick *Rhipicephalus sanguineus*	BHC, dieldrin	N. Jersey, Panama, Tex., Puerto Rico
African red tick *Rhipicephalus evertsi*	BHC, dieldrin	S. Africa
Brown ear tick *Rhipicephalus appendiculatus*	BHC, dieldrin	S. Africa
American dog tick *Dermacentor variabilis*	BHC, dieldrin	Mass.

Pest	Insecticides	Location
House fly *Musca domestica*	BHC, dieldrin	Calif., Sardinia, USA, Scandinavia, S. America, Africa, USSR, Japan, India, Caribbean, Romania
Stable fly *Stomoxys calcitrans*	BHC, dieldrin	Norway, Florida, Germany
Sheep blowfly *Phaenicia cuprina*	BHC, dieldrin	Norway, Florida, Germany, Australia
Green bottle fly *Phaenicia sericata*	BHC, dieldrin	New Zealand, S. Africa
African latrine *Chrysomyla putoria*	BHC, dieldrin	Congo, Malagasy, Zanzibar
Horn fly *Haematobia irritans*	BHC, dieldrin	Texas
Little house fly *Fannia canicularis*	BHC, dieldrin	Calif.
Fannia femoralis	BHC, dieldrin	Calif.
Midge *Chironmus zealandicus*	BHC, dieldrin	New Zealand
Midge *Glyptotendipes paripes*	BHC, dieldrin	Florida
Filter fly *Psychoda alternate*	BHC, dieldrin	England
Biting midge *Cullcoides furens*	BHC, dieldrin	Florida, Panama
Eye gnat *Hippelates collusor*	BHC, dieldrin	Calif.
Borborid fly *Leptocera hirtula*	BHC, dieldrin	Malaya

Pest	Insecticides	Location
Culex fatigans (quinquesfasciatus)	BHC, dieldrin	Calif., Malaya, India, E. Asia, S. America, W. Africa, Panama, Zanzibar, Congo, Tex., Mali, Madagascar, Brazil, Tanganyika, China, Togo, Ivory Coast, Queensland
Culex pipiens	BHC, dieldrin	Italy, Israel, France, Japan, Korea, Morocco
Culex tarsalis	BHC, dieldrin	Calif., Ore.
Culex tritaeniorhynchus	BHC, dieldrin	Dahomey, Ryukyus, Korea
Aedes aegypti	BHC, dieldrin	Puerto Rico, Jamaica, Haiti, Curacas, Virgin Islands, Surinam, Guyane, Cambodia, S. Vietnam, Tex., Cameroun, Tahiti, Thailand, Congo, Senegal, Ivory Coast, Liberia, Togo, Nigeria, Upper Volta
Aedes sollicitans	BHC, dieldrin	Florida, Delaware
Aedes taeniorhynchus	BHC, dieldrin	Florida, Georgia
Aedes nigromaculis	BHC, dieldrin	California
Aedes melanimon	BHC, dieldrin	California
Aedes cantator	BHC, dieldrin	New Brunswick
Psorophora confinnis	BHC, dieldrin	Mississippi
Psorophora discolor	BHC, dieldrin	Mississippi
Anopheles sacharovi	Dieldrin	Greece

Pest	Insecticides	Location
Anopheles quadrimaculatus	Dieldrin	Miss., Ga., Mex.,
Anopheles gambiae	Dieldrin	Nigeria, Liberia, Ivory Coast, Dahomey, Upper Volta, Cameroun, Sierra Leone, Togo, Ghana, Mali, Conga (Brazz), Sudan, Mauritius, Madagascar
Anopheles subpictus	Dieldrin	Java, Ceylon, N. India, W. Pakistan
Anopheles coustani	Dieldrin	Arabia
Anopheles pulcherrimus	Dieldrin	Arabia
Anopheles albimanus	Dieldrin	Salvador, Guatemala, Nicargua, Honduras, Jamaica, Ecuador, Mexico, Br. Honduras, Cuba, Dominican Rep. Haiti, Colombia
Anopheles pseudopunctipennis	Dieldrin	Mexico, Nicaragua, Peru, Venezuela, Ecuador
Anopheles aquasalis	Dieldrin	Trinidad, Venezuela, Brazil
Anopheles culcifacies	Dieldrin	W. India, Nepal
Anopheles vagus	Dieldrin	Java, Philippines
Anopheles barbirostris	Dieldrin	Java
Anopheles annularis	Dieldrin	Java
Anopheles sergenti	Dieldrin	Jordan
Anopheles fluviatilis	Dieldrin	Arabia
Anopheles splendidus	Dieldrin	N. India

Pest	Insecticides	Location
Anopheles stephensi	Dieldrin	Iran, Iraq
Anopheles minimus flavirostris	Dieldrin	Philippines, Java
Anopheles Pharoensis	Dieldrin	Egypt, Sudan, Israel
Anopheles albitarsis	Dieldrin	Colombia, Venezuela
Anopheles labranchiae	Dieldrin	Morocco, Algeria
Anopheles strodei	Dieldrin	Venezuela
Anopheles triannulatus	Dieldrin	Venezuela, Colombia
Anopheles sundaicus	Dieldrin	Java, Sumatra, Sabah
Anopheles aconitus	Dieldrin	Java, India
Anopheles neomaculipalpus	Dieldrin	Trinidad, Colombia
Anopheles crucians	Dieldrin	Carolina, Dominican Rep.
Anopheles filipinae	Dieldrin	Philippines
Anopheles maculipennis	Dieldrin	Romania
Anopheles rangeli	Dieldrin	Venezuela
Anopheles maculipennis messeae	Dieldrin	Romania
Anopheles labranchiae atroparvus	Dieldrin	Romania, Bulgaria
Anopheles philippinensis	Dieldrin	Sabah
Anopheles funestus	Dieldrin	Nigeria, Ghana, Kenya
Anopheles nili	Dieldrin	Ghana
Anopheles rufipes	Dieldrin	Mali

(a) Source (4).

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TOXAPHENE RESISTANCE IN ANIMALS
OTHER THAN INSECTS, MITES AND TICKS

Since many pest species have developed resistance to the chlorinated hydrocarbon insecticides, it seems unlikely that nontarget species have remained unaffected. That nontarget organisms have been affected is shown by the occurrence of small numbers of resistant individuals in susceptible populations of certain fish coupled with cross-resistance and retention of resistance in several generations of fish reared in the absence of insecticides. This suggests that a genetically based development of insecticide-resistant strains of fish have evolved in areas which have been subjected to intensive insecticidal treatment (1 and 13).

Endrin resistance in mosquito fish was attributed to physiological tolerance, when no evidence of excretion or detoxification was found (10 and 11). In heavily contaminated environments supporting insecticide resistant strains of marine organisms, top piscivores such as largemouth bass maybe absent. This suggests that selection in the food chain may occur through biological magnification. Presumably, the resistant fish which survive accumulate and tolerate high levels of residues. These individuals aggravate the problem, because the predators which feed on them may be killed by the insecticides in the bodies of the resistant fish.

Insecticide contamination of runoff water apparently is a major factor involved in the development of insecticide resistant fish populations (13). According to Ferguson et al (5 and 6), muds from natural waters in runoff from cotton fields may contain sorbed pesticides greatly in excess of levels lethal to certain fish. Although lethal quantities of these sorbed insecticides can be extracted with organic solvents, they are not released in lethal amounts into standing water.

Resistance in fish. Resistance and cross-resistance has been reported in populations of mosquito fish to DDT, endrin, aldrin, dieldrin, toxaphene and heptachlor (1 and 2). Although fish may be cross-resistant to an insecticide to which they have had no prior exposure, the nature of cross-resistance seems to differ from that of insects. Only low levels of DDT-resistance are known in fish; cross-resistance to DDT is poorly developed or absent.

Resistance in invertebrates other than insects. Ferguson et al (4, 5, 6) states that invertebrates known to contain resistant populations include a clam, Eupera singleyi; a snail, Physa gyrina; 6 species of cyclopoid copepods and a freshwater shrimp, Paleomonetes kadiakensis.

Resistance in vertebrates. Among the vertebrates, pesticide-resistance has been demonstrated in fishes, anuran amphibians and mammals. Six species of fishes (golden shiner, black bullhead, yellow bullhead, mosquito fish, bluegills, and green sunfish) from cotton-producing areas in the Mississippi delta are known to be resistant when compared with the same species for areas of minimal pesticide use. Most species resist several pesticides, particularly the chlorinated hydrocarbons endrin, toxaphene and Strobane (4, 8 and 15).

Northern and southern cricket frogs and Fowler's toads near cotton fields show as much as 50-fold levels of resistance when tested against several chlorinated hydrocarbon insecticides (10).

In Virginia apple orchards endrin did not control wild pine mice in certain areas. Webb and Horsfall (19) reported a 12-fold endrin resistance in the pine mouse, Pitymys pinetorum.

Ferguson (10, 11) indicated that in general, levels of resistance are highest for the most stable chlorinated hydrocarbons especially the cyclodiene derivatives including toxaphene.

Summary

Invertebrates other than insects known to be resistant to chlorinated hydrocarbon insecticides include a clam, a snail, a freshwater shrimp and 6 species of cyclopoid copepods.

Among the vertebrates, resistance has been demonstrated in fishes, anuran amphibians and a mammal. Six species of fish are resistant to insecticides in the cotton-producing areas in the Mississippi delta when compared with the same species collected from areas of minimal pesticide use.

Among the amphibians, northern and southern cricket frogs and Fowler's toad from near cotton fields show as much as 50-fold levels of resistance when tested against several chlorinated hydrocarbon insecticides.

A wild population of pine-mice is resistant to endrin.

SPECIES RESISTANT TO CHLORINATED HYDROCARBONS

Black bullhead
Ictalurus melas

Yellow bullhead
Ictalurus natalis

Golden shiner
Notemigonus crysoleucas

Mosquito fish
Gambusia affinis

Bluegills
Lepomis macrochirus

Green sunfish
Lepomis cyanellus

Cyclopoid copepods
- *Eucyclops agilis*
- *Orthocyclops modestus*
- *Macrocyclops albids*
- *Cyclops vernalis*
- *Cyclops bicuspidatus*
- *Cyclops varicans*

Pine mouse
Pitymys pinetorum

Clam
Eupera singleyi

Snail
Physa gyrina

Freshwater shrimp
Paleomonetes kadiakensis

Fowler's toad
Bufo woodhousei fowleri

Cricket frog
Acris crepitans

Cricket frog
Acris gryllus

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